THE BLACK FLIES (DIPTERA: SIMULIIDAE) OF FLORIDA AND THEIR INVOLVEMENT IN THE TRANSMISSION OF *Leucocytozoon smithi* TO TURKEYS

By

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TABLE OF CONTENTS

	Page
ACKNOWLEDGEMENTS	ii
LIST OF TABLES	vi
LIST OF FIGURES	vii
ABSTRACT	xiii
CHAPTER	
I. INTRODUCTION	1
II. LITERATURE REVIEW	3
Simuliidae	3
Taxonomy and Distribution	3
Bionomics	7
Damage	22
Control	23
Florida Ecological Habitats	27
Leucocytozoon smithi	33
III. MATERIALS AND METHODS	40
Black Fly Survey	40
Leucocytozoon smithi Transmission	45
IV. RESULTS AND DISCUSSION	54
Simuliidae	54
General Comments	54
Introduction to the Black Fly Keys	64
A key to the larvae of the black flies of Florida	71
A key to the pupae of the black flies of Florida	74

TABLE OF CONTENTS
(Continued)

Page

A key to the adult male black flies of Florida 76
A key to the adult female black flies of Florida 78

Introduction to the Individual Species Sections. . . 80
Cnephia (Cnephia) ornithophilia Davies, Peterson, and Wood 81
Simulium (Byssodon) meridionale Riley. 92
Simulium (Byssodon) slossonae Dyar and Shannon . . . 99
Simulium (Eusimulium) congareenarum (Dyar and Shannon) 117
Simulium (Phosterodoros) dixiense Stone and Snoddy . 128
Simulium (Phosterodoros) haysi Stone and Snoddy. . . 137
Simulium (Phosterodoros) jenningsi Malloch 143
Simulium (Phosterodoros) jonesi Stone and Snoddy . . 155
Simulium (Phosterodoros) lakei Snoddy. 169
Simulium (Phosterodoros) notiale Stone and Snoddy. . 181
Simulium (Phosterodoros) nyssa Stone and Snoddy. . . 189
Simulium (Phosterodoros) taxodium Snoddy and Beshear. 195
Simulium (Psilozia) vittatum Zetterstedt 206
Simulium (Simulium) decorum Walker 224
Simulium (Simulium) tuberosum (Lundström). 235
Simulium (Simulium) verecundum Stone and Jamnback. . 255
Cnephia species Undetermined No. 1 268
Simulium species Undetermined No. 1. 271

Leucocytozoon smithi Transmission 277

V. CONCLUSIONS 294

LITERATURE CITED. 296

APPENDIX. COLLECTION SITE NAMES AND LOCATIONS. 319

BIOGRAPHICAL SKETCH 331

LIST OF TABLES

Table		Page
1.	Florida black fly species.	55
2.	Florida black fly distribution records by county	57
3.	Black fly associations based on collections of immature stages	59
4.	Black flies captured in Manitoba traps	60
5.	Sentinel turkey locations and results.	278
6.	Black flies captured in ramp traps	281
7.	Blackout box trapping results.	282
8.	Black fly captures from exposed turkeys.	283
9.	*Leucocytozoon smithi* transmissions	289

LIST OF FIGURES

Figure — Page

1. A modified Manitoba trap with a black plastic skirt 44
2. Sentinel turkeys in an exposure cage. 47
3. A blackout box trap in the field. 47
4. One view of a ramp trap 48
5. An exposed turkey in the field. 48
6. Glass container and paper cartons used for holding black fly adults alive in the laboratory. 51
7. Locations in Florida where black flies have been collected 56
8. Seasonal occurrence of black flies in Florida 63
9. Dorsum of the head capsule of a black fly larva *(S. slossonae)*. 65
10. Venter of the head capsule of a black fly larva *(C. ornithophilia)* 65
11. Lateral view of two black fly larvae *(S. dixiense)* 66
12. Black fly pupa and cocoon *(S. dixiense)*. 66
13. A wing of the black fly *Cnephia ornithophilia* 68
14. A frontal view of the head of a female black fly *(S. notiale)*. 68
15. The male genitalia of a black fly, *Cnephia ornithophilia* 69
16. The distal portion of the hind leg of a *S. meridionale* female. 69
17. The terminalia of a female *S. meridionale* 70
18. The head spots of a larva of *C. ornithophilia* 82

LIST OF FIGURES
(Continued)

Figure		Page
19.	The pupal exuvium and cocoon of *C. ornithophilia*	82
20.	Tarsal claw of a female of *C. ornithophilia*	84
21.	Genital fork and terminalia of a *C. ornithophilia* female	84
22.	Collection locations for *C. ornithophilia* in Florida	88
23.	Site 119, Gum Creek, a stream inhabited by *C. ornithophilia*	89
24.	The pupa and cocoon of *S. meridionale*	94
25.	The scutum of a *S. meridionale* female	94
26.	Collection locations for *S. meridionale* in Florida	97
27.	Gular notch of a *S. slossonae* larva	101
28.	The pupa and cocoon of *S. slossonae*	101
29.	Terminalia of a male of *S. slossonae*	102
30.	Terminalia of a female of *S. slossonae*	102
31.	Collection locations for *S. slossonae* in Florida	105
32.	Site 43, Double Run Creek, where *S. slossonae* immatures were collected	107
33.	Cephalic apotome of a *S. congareenarum* larva	118
34.	Venter of the larval head capsule of *S. congareenarum*	118
35.	Pupa and cocoon of *S. congareenarum*	120
36.	Terminalia of a male of *S. congareenarum*	120
37.	Terminalia of a female of *S. congareenarum*	121
38.	Collection locations for *S. congareenarum* in Florida	124
39.	Site 216, Turkey Creek, a typical *S. congareenarum* stream	125
40.	Dorsal view of the head capsule of a *S. dixiense* larva	130
41.	Gular notch and hypostomium of a *S. dixiense* larva	130

LIST OF FIGURES
(Continued)

Figure Page

42. Male terminalia of *S. dixiense*. 132

43. Terminalia of a female of *S. dixiense* 132

44. Collection locations for *S. dixiense* in Florida 134

45. Site 74, Pine Barrens Creek, a stream inhabited by *S. dixiense* 135

46. Cephalic apotome of a *S. haysi* larva. 138

47. Gular notch of a *S. haysi* larva 138

48. Pupal exuvium and cocoon of *S. haysi*. 139

49. Location of the collection site for *S. haysi* in Florida . . 141

50. Site 195, Juniper Creek, where *S. haysi* was collected . . . 142

51. Cephalic apotome of a *S. jenningsi* larva. 145

52. Gular notch of a *S. jenningsi* larva 145

53. A pupa and cocoon of *S. jenningsi* 146

54. *S. jenningsi* male terminalia. 146

55. Genitalia of a female of *S. jenningsi* 147

56. Collection locations for *S. jenningsi* in Florida. 151

57. Site 141 at Gulf Hammock where *S. jenningsi* was collected . 152

58. Cephalic apotome of a *S. jonesi* larva 156

59. Gular notch of a *S. jonesi* larva. 156

60. Respiratory organ of a *S. jonesi* pupa 157

61. Terminalia of a male of *S. jonesi* 159

62. Genital fork and terminalia of a female of *S. jonesi*. . . . 159

63. Florida collection locations for *S. jonesi*. 161

64. Site 210, the Fenholloway River, a *S. jonesi* collection location. 162

LIST OF FIGURES
(Continued)

Figure | Page

65. Cephalic apotome of a *S. lakei* larva. 171

66. Gular notch and hypostomium of a *S. lakei* larva 171

67. Pupa and cocoon of *S. lakei* 172

68. Dorsal view of a male of *S. lakei* 172

69. Terminalia of a male of *S. lakei*. 173

70. Terminalia of a female of *S. lakei*. 173

71. Collection locations for *S. lakei* in Florida. 175

72. Site 135, Otter Creek, a collection site for *S. lakei* . . . 176

73. Head spots of a *S. notiale* larva. 183

74. Gular notch of a *S. notiale* larva 183

75. Pupal exuvium and cocoon of *S. notiale*. 184

76. Scutum of a male of *S. notiale*. 184

77. Terminalia of a male of *S. notiale*. 185

78. Terminalia of a female of *S. notiale*. 185

79. Collection locations for *S. notiale* in Florida. 187

80. Site 88 at Chattahoochee where *S. notiale* immatures were found. 188

81. *S. nyssa* pupa and cocoon. 191

82. Collection locations for *S. nyssa* in Florida. 193

83. Site 116, Blue Creek, where *S. nyssa* was collected. 194

84. Cephalic apotome and head spots of a *S. taxodium* larva. . . 197

85. Gular notch of a *S. taxodium* larva. 197

86. Pupal exuvium and cocoon of *S. taxodium* 198

87. Male terminalia of *S. taxodium*. 198

88. Female terminalia of *S. taxodium*. 199

LIST OF FIGURES
(Continued)

Figure | Page

89. Collection locations for *S. taxodium* in Florida 201

90. The Ichetucknee River, a *S. taxodium* collection site. . . . 203

91. Head spots of a *S. vittatum* larva 209

92. Gular notch of a *S. vittatum* larva. 209

93. *S. vittatum* pupa and cocoon 210

94. Terminalia of a *S. vittatum* male. 210

95. Scutum of a female of *S. vittatum* 211

96. Terminalia of a female of *S. vittatum* 211

97. Collection locations for *S. vittatum* in Florida 217

98. Site 167 at Crestview where *S. vittatum* was collected . . . 219

99. Head spots of a *S. decorum* larva. 226

100. Gular notch of a *S. decorum* larva 226

101. *S. decorum* pupa and cocoon. 227

102. Male terminalia of *S. decorum* 227

103. Female terminalia of *S. decorum* 228

104. Collection locations for *S. decorum* in Florida. 232

105. Shepard's Mill, Site 86, where *S. decorum* was collected . . 234

106. The cephalic apotome of a *S. tuberosum* larva. 237

107. The gular notch of a *S. tuberosum* larva 237

108. Pupa and cocoon of *S. tuberosum* 238

109. Male terminalia of *S. tuberosum* 238

110. Terminalia of a female of *S. tuberosum*. 239

111. Collection locations for *S. tuberosum* in Florida. 243

112. Site 56 in Clay County, a collection spot for *S. tuberosum*. 246

LIST OF FIGURES
(Continued)

Figure | Page

113. The head spots of a *S. verecundum* larva. 257

114. The gular notch of a *S. verecundum* larva 257

115. The pupa and cocoon of *S. verecundum* 258

116. Male terminalia of *S. verecundum* 258

117. Terminalia of a female of *S. verecundum*. 259

118. Collection locations for *S. verecundum* in Florida. 262

119. Panther Creek, Site 223, where *S. verecundum* was collected .264

120. Cephalic apotome of a larva of *Cnephia species* No. 1 269

121. Venter of the head capsule of a larva of *Cnephia species* No. 1. 269

122. Collection location for *Cnephia species* No. 1 in Florida . .270

123. Pupal exuvium and cocoon of *Simulium species* No. 1 273

124. Larval head spots of *Simulium species* No. 1. 274

125. Venter of the head capsule of the larva of *Simulium species* No. 1. 274

126. Collection location for *Simulium species* No. 1 in Florida. .275

127. Small flow to Pine Barrens Creek, Site 74, where *Simulium species* No. 1 was collected 276

128. Gametocytes (G) of *L. smithi* among normal turkey blood cells. 291

129. Ookinetes of *L. smithi* .291

130. Sporozoites of *L. smithi* photographed in saline. 292

131. Stained sporozoites of *L. smithi* 292

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THE BLACK FLIES (DIPTERA: SIMULIIDAE) OF FLORIDA AND THEIR
INVOLVEMENT IN THE TRANSMISSION OF
Leucocytozoon smithi TO TURKEYS

By

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Immature and adult black flies (Diptera: Simuliidae) were collected in Florida over a period of three years. Eighteen species of black flies including ten which are new records for the State were found to occur in Florida. Records of black flies from 192 locations in 50 counties are included. Biological information is provided for each species together with data on distribution, seasonal occurrence, stream ecology and species associations. Keys to the Simuliidae of Florida are provided and structures are illustrated. Representative specimens have been deposited in the Florida State Collection of Arthropods and in the United States National Museum, Washington D.C.

Transmission investigations have incriminated three species, *Simulium congareenarum*, *S. meridionale*, and *S. slossonae*, as vectors of *Leucocytozoon smithi* to turkeys in Florida. On nineteen occasions *L. smithi* was transmitted to domestic turkeys by the bites of infected black flies.

CHAPTER I
INTRODUCTION

Members of the family Simuliidae as immatures inhabit flowing water and as adults are often blood feeders which may vector diseases to man and animals in many parts of the world. A knowledge of the composition, distribution, ecology, and habits of the simuliid fauna of any area of local concern is essential for proper assessment of the impact on man of these insects. Successful black fly control is dependent on thorough knowledge of the major breeding sites, seasonal occurrence and other facts concerning local Simuliidae.

When I arrived in Florida to begin my Ph.D. research I wrote to the Florida Division of Health offices in Jacksonville, Vero Beach, and Panama City and enquired about previous investigations on the black flies of Florida. I found that little work had been done in the State on this family of biting flies (Beck, 1973; Linley, 1973; Rogers, 1973 — all personal communications). Mrs. A.T. Slosson collected black flies in Florida (Dyar and Shannon, 1927), probably at the turn of the twentieth century. Some of Mrs. Slosson's specimens and a portion of those of Calvin Jones, Harry Gouck, Darrell Anthony and other USDA researchers who collected black flies in the northern and central portions of the State in the 1940's and 1950's are located in the U.S. National Museum. Based at least in part on the examination of these specimens Stone (1965) listed the following seven species from Florida: *Cnephia (Cnephia) pecuarum* (Riley); *Simulium (Eusimulium)*

congareenarum(Dyar and Shannon); *Simulium (Byssodon) meridionale* Riley; *Simulium (Byssodon) slossonae* Dyar and Shannon; *Simulium (Simulium) decorum* Walker; *Simulium (Simulium) jenningsi* Malloch; and *Simulium (Simulium) tuberosum* (Lundström). Stone and Snoddy (1969) did not list *S. jenningsi* as occurring in Florida but did record two new species *Simulium (Phosterodoros) jonesi* Stone and Snoddy and *Simulium (Phosterodoros) nyssa* Stone and Snoddy from the State. Thus the total number of species of black flies known to occur in Florida when I began my research was eight.

This research was initiated in the fall of 1973 with the following objectives: 1) to determine the species complement, distribution and seasonal occurrence of black flies throughout Florida; 2) to gather ecological information primarily on the immature stages; 3) to provide keys with illustrations to the black flies of Florida; 4) to gather and deposit in the U.S. National Museum of Natural History and the Florida State Collection of Arthropods as complete a collection of adult and immature Florida black flies as possible; and 5) to determine the vectors of *Leucocytozoon smithi* in turkeys in Florida.

CHAPTER II
LITERATURE REVIEW

Simuliidae

Taxonomy and Distribution

Rubtsov (1974) presents a concise discussion on the history and major advances in black fly taxonomy. Linnaeus (1758) first described two simuliids and assigned the names *Culex reptans* and *Culex equinus* but did not differentiate them from mosquitoes (Davies et al., 1962). Latreille created the name *Simulium* for the genus in 1802 using *Rhagio colombaschensis* Fabricus as the type (Stone and Jamnback, 1955). This remained the only genus for the thirty to forty black fly species described up to the turn of the twentieth century. The work of Smith and Kilbourne (1893) and others which focused on arthropods as vectors of diseases stimulated much research on insects and related groups of medical and veterinary importance. Roubaud (1906) designated two black fly subgenera, *Prosimulium* and *Eusimulium*, based on differences in wing venation. Lundström (1911) in Sweden and Jobbins-Pomeroy (1916) in the United States were early taxonomists who made use of the male terminalia of simuliids to separate species. Edwards (1915) in England also used male terminalia but more significantly in 1920 stressed the importance of studying black fly larvae and pupae. Dyar and Shannon (1927) first used female genitalia to separate North American black flies. Enderlein (1930) divided the family Simuliidae into 29 genera. Smart (1945) took a more conservative approach and distinguished 6 genera in the family:

Parasimulium Malloch, *Prosimulium* Roubaud, *Austrosimulium* Tonnoir, *Gigantodax* Enderlein, *Cnephia* Edwards and *Simulium* Latreille. Stone (1963) listed 11 genera and 22 subgenera in the Simuliidae which he considered valid. Crosskey (1969) stressed a trinomial approach to simuliid taxonomy with heavy reliance on subgenera. Rubtsov (1974) recognized 17 genera of black flies in the Palaearctic region and listed 59 genera for the Simuliidae of the world, many of which American and British authors regard as subgenera.

Brues et al. (1954) provide a worldwide bibliography of important papers which deal with the taxonomy of the Simuliidae up to the early 1950's. In addition to the works already mentioned outstanding publications since about 1950 dealing with the classification and distribution of black flies outside of the United States include:

Palaearctic region — Davies (1968) - Britain, Carlsson (1969) - Spain, Rubtsov (1956 and 1962) - U.S.S.R., Kuusela (1971) - Finland, Zivkovic (1971) - Yugoslavia, Rivosecchi (1971 and 1972) - Italy, Crosskey and Peterson (1972), and Zwick (1974) - Germany;

Ethiopian region — Crosskey (1957) - West Africa, Travis et al. (1967), Crosskey (1969) - Africa and its islands, Fain and Elsen (1973) - Cameroons, Lewis and Raybould (1974) - Tanzania;

Oriental region — Travis and Labadan (1967a) - Asia and European U.S.S.R., Delfinado (1969, 1971) - Philippines, Crosskey (1973), Takaoka (1973) - Nansei Is., Uemoto et al. (1973) - Japan, Lewis (1973) - Pakistan, Datta (1973, 1974, 1975) - India;

Australian — Dumbleton (1963, 1972) - Australia and New Zealand, Travis et al. (1968);

Neotropical — Leon and Wygodzinsky (1953) - Ecuador, Dalmat (1955) -

Guatemala, Vulcano (1967), Travis and Labadan (1967b), Barreto (1969) - Colombia, Wygodzinsky and Coscaron (1970, 1973), Wygodzinsky and Najera (1970), Wygodzinsky (1971) - Northern Andes, Perez (1971) - Venezuela, Rubtsov and Avila (1972) - Cuba, Travis et al. (1974);

Nearctic (excluding the U.S.) — Syme and Davies (1958), Davies et al. (1962) - Ontario, Wood et al. (1963) - Ontario, Travis et al. (1969), Peterson (1970) - Canada and Alaska, Lewis and Bennett (1973) - Newfoundland.

In addition to the above works those of Rubtsov (1970, 1974) and Coscaron and Wygodzinsky (1973) mention the variability which has been observed in characters that are used for taxonomic determination of black flies. Gambarian and Terterian (1973) applied a numerical taxonomy approach and using 100 characters tried to separate simuliids in the *Eusimulium* group. The subgenus was found to be very homogeneous and creation of supraspecific taxa was not statistically justified. Internationally, black fly species determinations based on classical external morphological characters and behavioral differences are being supported by cytological, specifically chromosomal, studies. Rothfels (1956) provided an introduction and overview on analytical techniques for cytologically comparing black flies. Landau (1962), Dunbar (1969), and Vajime and Dunbar (1975) used chromosomal differences to separate forms or sibling species of a variety of simuliids including disease vectors where previously each had been considered a single species.

Taxonomic works covering the whole United States include: Coquillett (1898, 1902), Malloch (1914), Jobbins-Pomeroy (1916), Dyar and Shannon (1927), and Stone (1965). Other publications were more regional in

scope. Western and midwestern articles include: Twinn (1938), Smith and Lowe (1948) - California; Nicholson and Mickel (1950) - Minnesota; Stone (1952), Sommerman (1953) - Alaska; Wirth and Stone (1956) - California; Stone and DeFoliart (1959); Anderson and Dicke (1960) - Wisconsin; Stone and Boreham (1965), Hall (1972, 1974) - California; and Corredor (1975) - Washington.

Northeastern U.S. works include: Johannsen (1903), Leonard (1926), Metcalf (1932), DeFoliart (1951), Stone and Jamnback (1955), Jamnback and Stone (1957), Jamnback (1969), Pinkovsky (1970), and, reporting the unusual find of a South American black fly in the U.S. Wygodzinsky (1973) - New York; O'Kane (1926) - New Hampshire; Frost (1949) - Pennsylvania; Dimond and Hart (1953) - Rhode Is.; Sutherland and Darsie (1960a and b) - Delaware; Stone (1964) - Connecticut; Holbrook (1967) - Massachussetts; Eckhart and Snetsinger (1969) - Pennsylvania; and Crans and McCuiston (1970a) - New Jersey.

In the southeastern U.S. Tucker (1920) gave accounts of personal experiences with black flies and reported two species, *S. pecuarum* and *S. meridionale*, from Louisiana. Jones and Richey (1956) conducted a survey of the black flies of Jasper County, South Carolina, and discussed the biology, ecology and relationships to *Leucocytozoon* in turkeys of one *Cnephia (pecuarum)* and seven *Simulium* species (*congareenarum*, *decorum*, *jenningsi*, *slossonae*, *tuberosum*, *venustum* and undescribed species). Snow et al. (1958) reported on the ecology, habits and distribution of black flies occurring in the Tennessee River Basin and recorded species in the three genera: *Cnephia (mutata)*, *Prosimulium (hirtipes*, *magnum*, plus undescribed species*)*, and *Simulium (decorum*, *fibrinflatum*, *jenningsi* sp. group, *meridionale*, *pictipes*, *tuberosum*, *venustum*, *verecundum*,

vittatum and three undescribed species). Snoddy and Hays (1966) mentioned that eleven species of Simuliidae were captured at one location in Alabama using a New Jersey light trap modified for daylight use by removing the light and substituting, as an attractant, carbon dioxide gas dispensed at .45 kg (1 lb)/hr. Stone and Snoddy (1969) present distribution records, descriptions, and some biological information for 28 species of black flies discovered or expected to occur in Alabama. Garris et al. (1975) present the seasonal distribution of 7 species of *Simulium* collected from streams in Sumter Co., South Carolina, and mention observations of black flies feeding on turkeys and captured in a CO_2-baited trap. Snoddy and Beshear (1968), and Snoddy (1971, 1976) describe and give some facts on the biology of three new species (*S. taxodium*, *podostemi* and *lakei*) in the expanding species list of the former *S. jenningsi* group.

Bionomics

The eggs of black flies are dropped freely into the water or are attached to substrates in the flows of streams and rivers. The larvae which develop feed by filtering material from the current or by scraping organic matter off the substrate and, usually within a month, transform into pupae. From the pupa which is normally attached to rocks or vegetation beneath the surface of the stream an adult fly emerges, in a bubble of gas, rises to the surface and is immediately capable of flight. The adult females of most black fly species suck blood and both male and female flies obtain energy from natural sugar sources such as nectar and honeydew.

Eggs. Ussova (1961) describes most black fly eggs as between .24

and .33 mm long and irregularly triangular in shape with rounded corners; the eggs of some *Cnephia* species, however, are said to be larger and elongate elliptical. Golini and Davies (1975) found black fly eggs were .228 mm long and .139 mm wide. Davies and Peterson (1956) examined the eggs of four genera *(Gymnopais, Prosimulium, Cnephia, Simulium)* and found no external sculpturing. The eggs are light in color when just laid and darken as the embryos develop. Davies and Peterson (1956) found that the eggs of *Gymnopais* and *Prosimulium* were the largest and those of *Prosimulium* and *Cnephia* were the narrowest of the genera checked. Black fly eggs are often found attached to vegetation and rocks in a swift flowing stream or river or lying free on the stream bottom. Tarshis (1968) conducted experiments on and reviewed accounts in the literature of desiccation and the overwintering of black fly eggs and concluded, as Wu (1930) had done earlier, that only those eggs which are kept moist, by damp stones or underground springs, etc., in apparently dry stream bottoms and other situations are able to remain viable. Tarshis (1968) found that freezing eggs of a number of species at 0 to -70°C killed the embryos, while he was able to maintain moistened eggs alive for 424 days at 2-9°C. Kurtak (1974) found that eggs of *Simulium pictipes*, recovered while encased in ice from rock crevices along a stream, hatched in the lab after three days in 10°C flowing water. In northern areas eggs of many black fly species hatch when the water temperature reaches around 8°C (Carlsson, 1967). Raybould and Grunewald (1975) found eggs of the Kibwezi form of *S. damnosum* hatch at 20°C four days after oviposition. Tarshis (1968) indicates normal egg hatch for Maryland black fly eggs occurs within one to five days.

Larvae - general morphology and development. Black fly larvae are usually between 4.5 and 10 mm long when mature, club-shaped with a wider

posterior end, and possess a pair of multiple-rayed cephalic fans on the anterior end, an unsegmented ventral proleg in the thoracic region, and a circlet of anal hooks posteriorly. Dark cephalic head spots mark the origins of cephalic muscles. Tarshis (1968) found that larvae hatched from eggs in the lab after 7-38 days at 10°C and 1-5 days at 20-25°C. Larvae developed to pupae in 18-50 days at 10°C and 11-29 days at 20-25°C. Cameron (1922), Dalmat (1955) and Reisen (1975) found six larval instars in the black flies they studied. Johnson and Pengelly (1970) observed *S. rugglesi* to pass through seven larval instars and Fredeen (1975) mentioned a seventh and final instar for *S. arcticum*. Craig (1975) distinguished nine larval instars in Tahitian species of black flies.

Larval habitat. Larvae frequently attach by their posterior hooks onto a patch of silk secreted by their large, looped salivary glands. The silk is often applied to the same substrate on which the eggs are found. Larvae may drift a short distance tethered to the old substrate by a silk thread (Tarshis and Neil, 1970). A larva may also move in a looping, geometrid fashion from silk patch to silk patch alternately attaching its proleg then its anal disc (Dalmat, 1955). Elliot (1971) reported that black fly larvae can actively migrate upstream as well as passively downstream. Clean, smooth items free of algae or slime are preferred attachment substrates for the larvae and larvae are often located on such objects in stream sections where the current is increased by a partial obstruction (Dalmat, 1955; Carlsson, 1967). Larvae show positive phototaxis and colonize light substrates faster and more densely than dark substrates (Carlsson, 1967). Unusual attachment sites include the phoretic associations of black fly larvae on prawns, mayfly nymphs, crabs, and other arthopods reported by

Disney (1971, 1973, 1975) and the attachment of young larvae of *S. damnosum* to older larvae of the same species (Burton, 1971). Carlsson (1967) in Sweden found the greatest concentrations of larvae in flows 80 to 120 cm/sec although some species preferred 40 cm/sec currents. Cariaso (1962) in the Philippines found immature black flies concentrated where the water velocity was .5-.86 m/sec (1.63-2.83 ft/sec). Wu (1930) reported that simuliid larvae remained well established at a velocity of 1.83 m/sec (6 ft/sec). Rohdendorf (1974) suggested that the closed respiratory system of black fly larvae resulted after invasion of the swift flowing well oxygenated habitat typical of most simuliids. Tarshis (1968) found that black fly larvae could not live more than eight hours in still water. Anderson and Shemanchuk (1975), however, report they transported black fly larvae for several days in shallow, non-agitated, ice-chilled water with little mortality. Wu (1930) found approximately equal amounts of dissolved oxygen in quiet and turbulent sections of black fly streams and after a number of experiments concluded that black fly larvae have a definite requirement for current to lessen sedimentation and supply adequate food, not because of improved oxygen conditions. Crans and McCuiston (1970b) found larvae in permanent rivers and streams, temporary creeks and flowing roadside ditches. Dalmat (1955) and Lewis and Bennett (1975) reported finding a few live larvae in situations where the flow was so slow that silt and mud present prevented the larvae from anchoring to any fixed object. Van Someren (1944) reported *S. ruficorne* larvae from small pools and footprints in sandy river beds in Somaliland. Certain species are typically found in limited types of lotic habitats: *S. arcticum* in large rivers (Cameron, 1922), *S. pictipes* at the outflow of dams and on

flat bedrock (Snow et al., 1958) and *S. ochraceum* in very small streams about one meter (a few feet) wide and a few centimeters (a few inches) deep (Dalmat, 1955). Travis and Vargas (1970) found that in contrast to the clear mountain streams, the lower, slower Costa Rican streams most polluted with sewage and garbage had the greatest larval and pupal populations. Cariaso (1962) reported that black fly larvae were discouraged from breeding in water polluted with nitrogenous and human wastes. Carlsson (1967) indicates moderate pollution increases organic drift and is good for larvae but heavy pollution clogs the cephalic fans. Streams in Wisconsin carrying large amounts of eroded soil particles and other detritus were observed to be poor habitats for most black fly larvae due to feeding interference (Anderson and Dicke, 1960).

<u>Larvae - feeding structures, behavior, and food</u>. Davies (1974) and Craig (1974) describe the evolution and development of the important larval feeding structures in many species, the cephalic fans, and mention how these lateral palatal brushes which are well developed filtering devices in most black flies are apomorphically absent in later instars of *Gymnopais* and *Twinnia* and modified into raking structures in *Crozetia* species. Chance (1970) stated that particles from about 1 to 350 microns in diameter, the majority in the 10 to 100 micron range, were ingested by four filter feeding species she studied. Larvae were found to attach within 10 centimeters of the water surface, extend their bodies into the flow and alternately open and close the fans combing particles from the fans when they were closed into the cibarium where a bolus was formed. Mulla and Lacey (1976) report larvae, with their heads pointed downstream, attach to silk deposits with their anal hooks, rotate the body 90° to 180° and open the cephalic fans to strain matter

from the flow. Chance (1970) suggests that the horizontally operating mandibles, especially in non-filtering, grazing forms like *Twinnia*, scrape organic food off the substrate. Rubtsov (1974) discusses the outer sclerite of the labium - the submentum or hypostomum, an anteriorly serrate structure bearing setae which lies just cephalad of the gular notch - and mentions that it is a tactile organ, is important in producing the larval silk strands, and that it may serve to scrape food from the substrate. In species which lack fans the submental teeth sometimes acquire a spatulate shape.

Jobbins-Pomeroy (1916) reported *Euglena* and *Spirogyra* were important food items for black fly larvae but Cameron (1922) found diatoms formed the main components of the food. Anderson and Dicke (1960) found diatoms, other algae, and considerable inorganic material in the intestinal contents of black fly larvae. Carlsson (1967) listed as larval food bacteria, plankton, plant and animal parts, pollen and aerial fallout. Spring flooding leads to large outbreaks of black flies when rising water temperatures trigger synchronous hatching of many black fly eggs. The rising stream and river waters make more substrate available for larval attachment and increase organic debris and hence food available for the larvae. Bacteria filtered out by *S. underhilli* led to speculation of using simuliid larvae as indicators of pollution (Snoddy and Chipley, 1971). Reisen (1974) found that larvae remove bacteria and organic and inorganic particulate matter. Young larvae of *Simulium* species in nature fed at a faster rate than did older larvae as indicated by the time necessary to void a plug of dye particles (Mulla and Lacey, 1976). These authors also found that at a lower temperature (12.8°C = 55°F) a longer time (35-55 min) was necessary to

eliminate a particulate plug from larval guts than at a higher temperature (30°C = 86°F, 20-30 min).

Pupae. Pupae attach to the same substrates as the larvae. The larvae form silken cocoons in which pupation occurs. Hinton (1958) mentions a pharate pupa stage where pupal characters are visible within the last larval skin but the larval mouth parts are still articulated and feeding continues up to when this "pupa" begins spinning the cocoon. The cocoons may be thin and rudimentary *(Twinnia)*, shapeless, irregular masses of silk *(Prosimulium* and some *Cnephia)*, sturdy, coarse cocoons *(Simulium pictipes)* or tapered, slipper-like finely shaped and tightly woven cocoons *(most Simulium)* (Stone and Jamnback, 1955). Members of the *Simulium* subgenus *Phosterodoros* have cocoons with forward edges that are convex in profile and have a large opening anteriorly on each side of the cocoon (Stone and Snoddy, 1969). Field and Low (1961) describe what Underhill (1944) illustrated for *S. jenningsi (*as *S. nigroparvum)*, that is, sexual dimorphism in the cephalic plate of simuliid pupae. The cephalic plate, a sclerite that begins between the bases of the antennal sheaths and extends over the cephalic area, is longer and narrower in the male than it is in the female pupa. The pupae usually face downstream and are bordered on each side by tubular filaments which vary in number and shape according to species and aid in respiration. Cameron (1922) observed that the number of pupal filaments in *S. arcticum* varied from the usual 12 to 13 or 11 and that one set might vary while the other group on the same pupa might bear the more typical number of filaments. Coscaron and Wygodzinsky (1973) mention the variation in the point of bifurcation or petiole lengths of the respiratory filaments of different specimens within the same species.

Adults - emergence. Tarshis (1968) reported that pupal development of three species of *Simulium* took 1 to 5 days at 20-25°C and that at 10°C it took 21 days following pupation before adults began emerging. Davies et al. (1962) describe how, at emergence, after gas fills the skin of the submerged pupa, the adult ruptures the pupal exuvium, pulls itself through a T-shaped opening and quickly floats to the surface in a bubble of gas (also see Hannay and Bond, 1971, section below). Carlsson (1967) found that a heavy silt cover on pupae prevented adults from emerging. Adults may easily be reared from pupae placed on moist filter paper in petri dishes (Hannay and Bond, 1971; Sutcliffe and McIver, 1974). Wenk (1965) found males emerge sooner from pupae than do females. Disney (1969) found three species of *Simulium* in Africa pupate by day and may prolong pupation to avoid emerging at night. On warm days eclosion occurred early in the morning, on cooler days eclosion peaks occurred during the late morning and mid-day and under artificially cold days emergence was shifted to the late afternoon.

Adults - appearance and morphology. Adult black flies rarely exceed 5 mm in length (Dalmat, 1955). Male and female black flies are hump-backed in appearance. The males are holoptic, usually darker and more velvety than females and often bear a shiny pair of anterior lateral spots on the scutum (Davies et al., 1962). Costalization (i.e., a strengthening of the anterior wing veins and a decrease in wing venation in the posterior portion of the wing), shortening of the legs, evolution of the calcipala and pedisulcus, increase in head size and shortening of the abdomen with reduction of tergites and sternites are considered apomorphic developments (Rohdendorf, 1974; Rubtsov, 1974). Hungerford (1914) in discussing the anatomy of *Simulium vittatum* adults noted that

black fly females possess a single spermatheca. Lewis (1957) presented morphological information about adults of *S. damnosum* and suggestions on dissection techniques. Bennett (1963b) found the shape and appearance of the salivary glands of adults valuable in differentiating species. Hannay and Bond (1971) found raised cylindrical buttons, each with a wax filament, between the macrotrichia on black fly wings and suggest these structures may aid the adult in keeping its wings dry during emergence. Sutcliffe and McIver (1974) described cleaning hairs and combs of cuticular teeth on the metathoracic legs and cleaning hairs on the prothoracic legs which are used to clean the wings and head appendages, respectively.

Adults - attraction and trapping. Wirth and Stone (1956) indicate that male black flies are readily collected at light traps. Snow et al. (1958) captured adults at light, on vegetation, in cars and biting mammalian hosts. Fallis and Smith (1964) succeeded in using ether extracts of birds as attractants for simuliids. Anderson and DeFoliart (1961) used a variety of caged birds to attract ornithophilic black flies to traps in Wisconsin. Golini and Davies (1971) found that females of *S. venustum* fly upwind to a carbon dioxide source and cease flying upwind if the CO_2 source is turned off. Snoddy and Hays (1966) and DeFoliart and Morris (1967) made use of the attractancy of CO_2 to capture simuliids. The silhouette of a target rather than the reflectance is more important in attracting black flies (Gillies, 1974). Peschken and Thorsteinson (1965) state that black flies are more attracted to stationary targets than to moving targets and simuliids show less discrimination between form and shape of three dimensional objects than some other hematophagous insects. Bradbury and Bennett (1974a)

found bloodseeking black flies in the genera *Prosimulium*, *Cnephia*, and *Simulium* were more attracted to black, blue, and red than white or yellow, especially when the colors were of low visible reflectance. Thorsteinson et al. (1965) describe a tripod canopy-type Manitoba fly trap which uses a large black sphere target to make the trap visually attractive to bloodsucking diptera. Bradbury and Bennett (1974b) found that black flies could distinguish between targets based on their color, largely independent of carbon dioxide flow rates. Carlsson (1967) stated lakes, shoals, bogs and swamps act as "collecting mirrors" for female black flies seeking ovipositioning spots. Female black flies seeking oviposition sites oviposited more on green and yellow colored plastic strips in a stream than on red, purple, white or black strips (Golini and Davies, 1975). The Malaise trap, a final valuable collecting device for black flies and other insects, was conceived as a passive or random flight interception trap; however there is some evidence that the degree of color contrast between the trap and background vegetation is an attractance factor (Roberts, 1970 and 1972).

Adult - mating. Females of *C. dacotensis* have mature eggs at the end of the pupal stage and copulate with males on damp rocks soon after emergence. In general, however, simuliids can mate any time from emergence to oviposition time (Davies and Peterson, 1956). Davies and Peterson (1956) were unable to induce male black flies to mate by broadcasting the sound of the female wing beat frequency. Compared to males which do not form mating swarms, males with mating flights have larger eyes and enlarged upper facets. Snow et al. (1958) observed that males of *S. pictipes* flew upside down with their abdomens curved upward, contacted females as they flew by overhead, settled to a solid substrate,

and copulated venter to venter with the female situated uppermost. The presence of about one male for every two hundred females of *Cnephia mutata* is considered evidence for parthenogenic reproduction (Davies and Peterson, 1956). Downes (1965) reports *Prosimulium ursinum* is a maleless species in which the females do not even emerge from the pupa but disintegrate shedding the eggs into the stream.

Adult - feeding. Black flies have been collected from flowers (Davies and Peterson, 1956). Hocking (1953) stated that black flies and other northern biting flies obtain the energy for flight almost entirely from floral nectar. Lewis and Domoney (1966) reviewed the importance of sugar feeding on bloodsucking, autogeny, and parasite development and reported finding glucose, sucrose, fructose and other sugars in the crops of 101 wild caught *Simulium*. There are scattered reports of black flies being attracted to and feeding on cold-blooded animals (Hagen, 1883; Jobbins-Pomeroy, 1916; Smith, 1969), but of more importance are their ornithophilic, mammalophilic, and anthropophilic bloodsucking habits. *Cnephia dacotensis*, *Gymnopais holopticus*, *Twinnia tibblesi* and a few other black fly species acquire sufficient nutrients during the larval stage to produce eggs and do not suck blood (Davies and Peterson, 1956; Shewell, 1957). Rohdendorf (1974) suggested that limited larval food stimulated the adult female black flies to hunt and feed on vertebrates. Cameron (1922) mentions the tenacity and persistence black flies exhibit when feeding and states that once the mouth parts are securely inserted into the skin the insect is not easily disturbed. Ussova (1961) states that the mandibles pierce a host's skin, the maxillae with recurved teeth anchor the proboscis in the skin, and alternating actions of the cibarial and pharyngeal pumps suck the blood into the esophagus.

James and Harwood (1969) indicate simuliids are telmophages or pool feeders which lacerate blood vessels with the toothed, transversely operating mandibles and vertically operating maxillae. Davies and Peterson (1956) record a wide, natural host range for some species such as *S. venustum* - duck, crow, heron, deer, and human. Others such as *P. hirtipes* (now three species) and *S. decorum* which feed on mammals in the field fed on birds when placed in vials on the avian species in the lab. Yang and Davies (1974) examined the salivary glands of three black flies and found an anticoagulant factor which keeps the blood fluid for movement into the gut. These authors also report an agglutin factor in flies at least twelve hours old.

Adult - egg development and egg laying. Cameron (1922) found development of the ovaries of female black flies followed a successful blood meal. Davies and Peterson (1956) describe females of several species with weak teeth or only hairs on the mandibles and maxillae. These flies do not suck blood and emerge with already mature eggs. Davies and Peterson (1956) indicate eggs develop five to twenty-one days after emergence with the longer time involving a prolonged blood meal search under natural conditions. Immature and mature eggs are found together in the ovaries which suggests at least two ovarian cycles (Cameron, 1922). Females may survive long enough for three batches of eggs but few probably live this long in the field (Davies and Peterson, 1956). Cameron (1922) reported that the eggs of *Simulium simile (=S. arcticum)* are oviposited on rocks in large cake-like masses embedded in a soft gelatinous matrix. Davies and Peterson (1956) mention *Prosimulium* and *Cnephia* species which deposit eggs freely while flying down or across a stream. Stone and Jamnback (1955) indicate *S. vittatum* lays strings

of eggs in a gelatinous matrix. Golini and Davies (1975) in Canada found that female black flies settled at the water line on trailing cattail leaves and deposited large (16 cm x 2 cm) irregular egg masses one to five layers deep in a gelatinous substance. Davies and Peterson (1956) found simuliids oviposited an average of 200 to 500 eggs per female while Golini and Davies (1975) report an average of 417 eggs for each female.

Adults - life cycle, longevity, range, and general activity. Tarshis (1968) reports a 21 to 25 day period for mixed cultures of five *Simulium* species to develop into adults after eggs were placed in aquaria. Raybould and Grunewald (1975) found developmental time from egg to adult ranged from 18 to 50 days for African black flies.

Dalmat (1955) in Guatemala used colored dyes and the release-recapture technique and found marked female black flies survived as long as 85 days and traveled as far as 9.7 miles. Bennett (1963a) tagged females of *S. rugglesi* by feeding them on ducks injected with a phosphorous-32 solution and recovered labelled flies up to 28 days later and 9.6 km distant following watercourse paths. West et al. (1968) exposed black fly larvae and pupae to ^{32}P-treated water and recovered radioactive flies as far as 33.5 km (20.8 mi) distant. Bennett and Fallis (1971) found *S. euryadminiculum* flew up to five miles from the release point and report the average life span of the females was at least two to three weeks. Hocking (1953) summarized published records of flight ranges which reached a maximum of 145 km for *S. reptans columbaczense*.

Wellington (1974) reported frenzied activity in black flies which was apparently correlated with barometric pressure changes as a storm system approached. Cameron (1922) observed swarms of male black flies

or males and females on warm, cloudy days with rain threatening or falling gently. Carlsson (1967) reports that rapid changes in temperature, air pressure, and light seem to increase the activity of all simuliid species but he also mentions even a light breeze reduced black fly activity considerably. Hocking (1953) found flight was continuous in *Simulium* species down as low as 12.8°C (55°F).

Concerning nocturnal behavior Dalmat (1955) observed that black flies in Guatemala move down to the base of plants as the sun sets and can be captured at night using a lantern and sheet when the insects emerge from their resting sites near the ground. Wolfe and Peterson (1960) describe climbing trees and sweep netting twenty-five feet off the ground to capture black flies at night in Quebec. At dawn Wolfe and Peterson observed simuliids flying down from the canopy. Recently in India researchers used light traps and found that black flies were active throughout the night with a peak of flight activity occurring around midnight (Datta and Dasgupta, 1974).

Lab colonization. Black flies have been collected in nature in all stages and have been brought to the lab for experiments. Tarshis (1965a) describes techniques for collecting and shipping viable black fly eggs. Tarshis (1965b) and Tarshis and Adkins (1971) discuss collecting large numbers of black fly larvae on artificial cloth substrates and mention techniques for transporting larvae in aerated containers. Tarshis (1971) discusses rearing black fly pupae to adults in individual brass strainer cloth cylinders connected to aquarium air stone units. Tarshis (1972, 1973) describes field collection, laboratory rearing of immatures to adults, and successful feedings of females of *C. ornithophilia* on ducklings in the lab.

Wenk and Raybould (1972) point out that colonies of insects aid critical studies on their biology, provide adults free from infection for transmission work and can provide abundant material for investigations at times when natural populations are limited. Dunbar (1969) mentions that hybridization experiments supported by colonies of flies and sufficient knowledge of mating requirements can provide valuable information concerning the species status of insect forms. Dalmat (1955) found carbon dioxide stimulated feeding and oviposition and succeeded in inducing 40% of 65,000 *S. ochraceum*, *S. callidum*, and *S. metallicum* to feed and 20% to oviposit but no eggs developed into larvae. Field et al. (1967) found that females of *S. vittatum* confined in a vial bit man, quail, and rabbits. By impaling a male on a minuten and brushing the male genitalia against the genitalia of the female coupling could be achieved. No female of *S. vittatum* which mated survived to oviposit and eggs deposited by other, unmated, females in the artificial flows available failed to hatch. In the laboratory Wenk (1965) successfully reared, mated, blood fed and achieved oviposition of viable eggs with *Boophthora erythrocephala*. Wenk and Raybould (1972) working with the Kibwezi form of *S. damnosum* likewise reared, mated, blood fed and obtained viable eggs from female black flies in the lab. Mating, which was also achieved with a member of the *S. bovis* complex, occurred in partially lighted emergence cages and was confirmed by the presence of sperm or spermatophores. Raybould and Grunewald (1975) review the literature on lab colonization of black flies and mention difficulties confronting researchers: inducing mating, inducing females to feed on blood sufficiently and consistently, erratic viability or hatchibility of eggs oviposited in the lab, and removing wastes from immature rearing setups.

Although a few black fly species have been induced to complete every stage of their life cycle in the lab, they are still not considered to be successfully colonized.

Damage

Jamnback (1973) describes the black fly bite reaction: a hemorrhagic spot which develops into an itching wheal; sensitive individuals may suffer headache, fever, nausea, glandular enlargement, and bites around the eyes may cause swelling that results in obscured vision. Loomis et al. (1975) report black flies cause considerable irritation and tissue damage to horses' ears, head, neck, and belly, producing wounds and papules. *S. arcticum*, *C. pecuarum*, and *S. columbaczense* are described as major pests of livestock (Jamnback, 1973). Fredeen (1974) mentions black fly outbreaks during 1944-1947 in Saskatchewan which resulted in the deaths of 1100 farm animals including cows, horses, hogs, and shorn sheep. Fredeen (1975) reports a *S. arcticum* outbreak in 1972 along the Northern Saskatchewan River that killed at least 18 cattle. Black flies transmit *Leucocytozoon* parasites to ducks (Fallis and Bennett, 1966), trypanosomes to ducks (Desser et al., 1975), and *Leucocytozoon* parasites to turkeys (Skidmore, 1931; Noblet et al., 1972) (see also the *Leucocytozoon smithi* sections in the Literature Review and Results and Discussion below). Sudia et al. (1975) mention that epidemic VEE virus has been isolated from *Simulium* species in Colombia and although biologic transmission has not been proven, mechanical transmission by contaminated mouth parts may be possible for at least 72 hours. Travis et al. (1974) report a finding of vesicular stomatitis virus in black flies in Colombia. Eastern encephalitis virus has been

isolated from a pool of 100 unengorged *S. meridionale* which indicates this species may biologically transmit the disease (Anderson et al., 1961).

Steelman (1976) reports monetary losses in cattle herds due to worm nodules caused by simuliid transmitted *Onchocerca gutterosa* amount to $500,000 each year in Australia. *Onchocerca volvulus*, the causative agent of blinding filariasis in man, is vectored by black flies and has been reported from Africa, Yemen, Mexico, Guatemala, Venezuela, Colombia, and Surinam; indigenous cases may also occur in Brazil (World Health Organization, 1971; Travis et al., 1974; Raybould and Grunewald, 1975). Microfilaria of *O. volvulus* previously were observed in the eyes and skin, however, Anderson et al. (1975a) found microfilaria in the urine, blood, and sputum of persons treated with diethylcarbamazine. Diurnal periodicity of *O. volvulus* microfilaria in the skin has been shown to correspond with the peak feeding periods of important black fly vectors in Guatemala and Africa (Anderson et al., 1975b). Duke et al. (1975) found the transmission potentials of *O. volvulus* along breeding rivers for *S. damnosum* in Africa so high that no communities could survive there.

Control

Physical control. Impoundage of rivers and the creation of reservoirs together with planned periodic cutoffs of discharge water has discouraged or eliminated black fly breeding along many stretches of rivers (Snow et al., 1958). Removing debris such as planks, tree limbs, trailing vegetation and other potential substrates for black fly larvae can help control simuliid population levels in streams (Jamnback, 1973).

Chemical control. Carestia et al. (1974) reported that aerial applications of malathion, Dibrom[R], or Dibrom plus a repellent for adult black fly control were unsatisfactory. High winds, dropping temperatures and decreasing daylight interfered with adult fly activity and chemical effectiveness. Rapid reinfestation of adults from areas just outside the treated zone is another reason why most control efforts have been aimed at immature rather than adult black flies.

Fairchild and Barreda (1945) observed the effectiveness of DDT as a black fly larvicide. Davis et al. (1957) discussed early attempts at evaluating the larvicidal properties of parathion dripped into streams, dieldrin applied by air, and DDT delivered by various ground techniques as well as by airplane. Evidence of the persistence of DDT in non-target organisms and decreased susceptibility of black fly larvae from streams with a history of DDT treatments stimulated a switch to less persistent materials (Fredeen et al., 1971; Jamnback and West, 1970). Travis et al. (1970) in field trough tests found Dibrom was outstanding in producing mortality of *S. pictipes* larvae at .5 ppm and 1 ppm concentrations and stated Dibrom, Gardona[R] and Ciodrin[R] deserved more practical stream tests. Jamnback and Frempong-Boadu (1966) found chemical formulations are most effective which give uniform distribution of the insecticide in the water and have a specific gravity of slightly less than 1.0 to keep the toxicant near the surface where most of the black fly larvae are located. Abate[R] (20%) in Panasol plus .5% Triton X-161 (.998 specific gravity) applied by aircraft eliminated larvae up to .8 km (.5 mi) downstream from the treatment point. Pelsue et al. (1970) chose Abate as a black fly larvicide due to its low mammalian, avian, and fish toxicity and achieved 100% control of larvae in three locations

with .5 ppm delivered for 60 minutes. Detachment of larvae occurred within 24 hours and reinfestation was noted in 15 to 60 days. Kissam et al. (1975) reported 2% Abate Celatom granules delivered at 91 g AI/ .4 ha monthly reduced larval populations to zero and the populations only slowly built back up during the two weeks following each application.

Fredeen (1974) mentions that methoxychlor, another black fly larvicide, is minimally toxic to vertebrates and methoxychlor and its metabolites are not concentrated in fish or other aquatic species. In Canada in 1969 a 15 minute injection of .2 ppm methoxychlor emulsifiable concentrate caused the disappearance of 96% of the *S. arcticum* larvae 32 km downstream. Fredeen (1975) reported that a 7.5 minute injection of methoxychlor at .6 ppm killed 100% of the older black fly instars 40 and 80 km below the treatment area. Younger black fly instars which are more susceptible were depleted by 96% at a site 161 km downstream from the injection. Fredeen et al. (1975) indicate that methoxychlor adsorbs to suspended solids in the water and may act selectively against the filter-feeding black fly larvae. Following a methoxychlor application Wallace et al. (1973) noted increased drift but no eradication of non-target organisms and Fredeen (1975) found that non-target organisms repopulated more densely than before the treatment.

Investigations of the effect of insect developmental inhibitors on black flies indicate that significant reduction (75-100%) of adult emergence can be attained in the lab with at least three black fly species with Altosid[R] at concentrations between .001 and 1 ppm (McKague and Wood, 1974; Dove and McKague, 1975). Frommer et al. (1975) reported that in field evaluation tests in New York DEET-treated light mesh

jackets were effective in reducing landings per five minutes from 404 on controls to 1.2 on personnel with treated jackets.

Biological control. Cameron (1922) found that a fish of no food value called the common sucker fed on *S. simile (=S. arcticum)* larvae and pupae. Peterson and Davies (1960) review the predators of black flies which include adult Empididae and Dolichopodidae, Tendipedidae larvae, Trichoptera, Formicidae, Odonata - adults and naiads, and spiders. Sommerman (1962) reported empidid larvae fed on black fly larvae. Snoddy (1968) found the solitary wasp *Oxybelus emarginatum* to be a predator of adult black flies. Peterson and Davies (1960) and Chutter (1972) report cannibalism among simuliid larvae. Carlsson (1967) indicates predators are unlikely to serve well as man-manipulated biological control agents though the tricopteran genera *Hydropsyche* and *Rhyacophila* might give good results.

One of the earliest accounts of parasitism of black flies is that of Strickland (1911) who reported a mermithid nematode that retarded the development of the larval histoblasts and a sporozoan (microsporidian) which caused distorted and swollen larvae. Jamnback (1973) states that eighteen microsporidia have been described from black flies. Spores are ingested by the larvae, sporoplasm invades suitable host cells which are often in the fat bodies, multiplication and the production of many spores occurs, and the host larvae commonly die. Black fly pupae and adults have been found infected and transovarian transmission is common. Jamnback (1973) also mentions a fungus *Coelomycidium simulii* which occurs in black fly larvae and is usually fatal.

Davies (1958) reports microsporidians, mites, and mermithid nematodes as parasites of Canadian black flies and mentions that mermithids

are found in larvae, pupae and both sexes of the adults. From 15 to 60% of the females in emergence collections were found to be infected with mermithids and females in oviposition swarms were also found with mermithids. The infected females which attempt to oviposit may serve to disperse the mermithids and introduce them to additional stream environments. Welch (1964) indicated that the mermithid life cycle began with the consumption by black fly larvae of the mermithid, usually in the infective first juvenile stage. Molloy and Jamnback (1975) observed direct penetration of the black fly larval cuticle by preparasitic juveniles of *Neomesomermis fluminalis*, a mermithid which infects at least fourteen species of black flies in three genera. Exit of the parasite from the host is almost immediately fatal to the black fly. Anderson and DeFoliart (1962) report 49-93% parasitism by *Isomermis* and *Gastromermis* in one black fly species in Wisconsin. As a step toward the mass rearing of these biological control agents Bailey et al. (1974) discuss techniques for the mass collection of mermithid postparasites from field collected black fly larvae.

Batson et al. (1976) reported an iridescent virus from black fly larvae in Wales but could associate no major fluctuations in the black fly population with the presence of the infected larvae.

Florida Ecological Habitats

Florida is a peninsula that lies between 80 and 88 degrees west longitude and in the same latitude belt as the Sahara, Arabian, and other large deserts. Exclusive of the Keys, the State extends roughly 644 km (400 mi) north and south along the peninsula and about the same distance east to west along the north coast of the Gulf of Mexico. Florida covers

151,710 sq km (58,560 sq mi). The highest point in the State is 105 m (345 ft) above sea level, at Lakewood in Walton County. From Orlando south to Sebring another high section occurs and Iron Mountain near Lake Wales is located about 91.5 m (300 ft) above sea level (Morris, 1973). The earliest geological horizon is the Ocala limestone formed during the Eocene, 58 million years ago. Modern Florida made its first appearance during the Oligocene, 36 million years ago, as a small island about where the counties of Suwanee, Columbia, and Alachua are now located and then rose as a peninsula during the Miocene (Byers, 1930).

There are three schemes which have been used to break Florida up into sections according to: 1) vegetation; 2) geology; and 3) edaphic factors. These schemes are presented to help illustrate the ecological diversity in Florida, which on the one hand provides a range of habitats for simuliid species and on the other hand restricts black fly breeding to certain more suitable regions of the state. Byers (1930) divided Florida into seven biotic areas based on dominant types of vegetation: a tropical hammock strip from St. Lucie County through Dade County along the east coast extending a few miles inland with cabbage palms, mahogany, ironwood, papaya, epiphytic bromeliads, plus other plants; a grass swamp area south and east of Lake Okeechobee; magnolia and temperate hammocks, primarily along the east coast from Flagler County to Indian River County and on the Gulf Coast from Wakulla through Hernando Counties — these occur on well drained but moisture holding soils with magnolia, holly, and red bay as dominant vegetation on the east coast and beech, elm, sweetgum, hickory, and oaks dominant in the west coast hammocks; a section of southeastern deciduous forest extending down into Leon, Liberty and Jefferson Counties; tree swamps in Collier, Columbia, Baker

and Nassau Counties which include Big Cypress Swamp and other cypress and also black gum swamps; pine flatwoods along the Gulf west of Wakulla County, on the west coast of the peninsula from Pasco to Collier Counties, and in the Clay County and Putnam County region — these flatwoods occur on level, poorly drained land underlain by hard pan which results in an acid soil upon which grow palmetto, grass and pines; and, lastly, the southeast coniferous forest with long leaf, yellow, and slash pines and saw palmetto on well drained uplands.

Cooke(1939) divided Florida into five physiographic areas: central highlands, Tallahassee hills, western highlands, Marianna lowlands and coastal lowlands. The central highlands, in the center of the peninsula from Baker County and Columbia County south to Lake Okeechobee, contain in their southern section thousands of lakes which in the north section are accompanied by numerous streams. The Tallahassee hills and western highlands are composed mainly of red clay with a relatively good number of streams. The coastal lowlands usually lie less than 30 m (100 ft) above sea level and include swampy areas such as the Florida Everglades and marshy areas along the east and west coasts of the state. The Marianna lowlands in Walton, Holmes, Washington and Jackson Counties contain few permanent flows and consist mainly of swamps, flatwoods, and sandy hills bearing pines.

Davis (1967) and Smith et al. (1967) present six major land resource areas for Florida: a southern coastal plain; gulf coast flatwoods; central Florida ridge; Atlantic coast flatwoods; southern Florida flatwoods; and Everglades and associated areas. The southern coastal plain, occupying the northern half of the state from Escambia County to Madison County, is covered by forests of mixed hardwoods and pines on the lower

areas and forests of long leaf pines and oaks on upland clay or well drained upland sand. The gulf coast flatwoods — a strip along the gulf side of the panhandle from Escambia County through Citrus County, the Atlantic coast flatwoods primarily in Columbia, Baker, Nassau and Bradford Counties in the north, and the south Florida flatwoods occupying most of the southern section of the peninsula outside of the Everglades consist of pine flatwoods and swamp forests (pines, palmetto, herbs, bay tree, laurel, gum, cypress) on poorly to very poorly drained and marshy soils. The central Florida ridge is located on well drained soil in the middle of the peninsula and includes forests of long leaf pine and xerophytic turkey oak (now mainly planted in citrus) and hardwood forests of mixed evergreen and deciduous hardwoods on rich upland soils. The Everglades are primarily sawgrass, *Mariscus jamaicensis*, on peat and muck soils.

Average January temperatures range from 10-13°C (50-55°F) in the panhandle to 18-21°C (65-70°F) in the Everglades area. Average July temperatures fall between 27-29°C (80-84°F) throughout the state. Annual rainfall averages 134.62 cm (53 in) (Raisz, 1964). June through September or October is considered the rainy season in Florida (Byers, 1930) however Berner (1950) indicates that in the northwestern area rainfall is more evenly distributed throughout the year. There are seventeen first magnitude springs in Florida (Morris, 1973). Despite the heavy precipitation there are relatively few surface flows since much of the water moves in underground channels in the underlying limestone (Raisz, 1964). The vast majority of true streams and rivers are found north of Lake Okeechobee. Morris (1973) indicates there are 1,711 streams, rivers, and creeks in Florida with a total length of 16,989 km (10,550 mi).

Berner (1950) states that in Florida there are relatively few intermittent streams. He divides permanently flowing streams in the State into: sand-bottom creeks with loose shifting sand and little vegetation; sand-bottom creeks with fairly loose sand and considerable vegetation; silt-bottomed creeks with little vegetation; and silt-bottomed creeks with considerable vegetation. Stagnant rivers occur in south Florida where they serve primarily as drainage canals for the Everglades and though there is considerable vegetation along the shores of the rivers their fifteen to twenty-foot depths restrict plant growth toward the middle. Larger calcareous streams contain water which rises from springs, are basic in pH reaction, and in shallower sections are lined on the bottom with *Vallisneria* (eel grass), *Sagittaria* (arrowhead), algae and other plants. Erving (1971) notes that the temperature for Florida springs is about 22°C (72°F) year round. While noting springs are alkaline Berner (1950) also indicates that marsh and swamp water can be very acidic reaching a pH of 3.6 or below. He further adds slow flowing rivers to his classification of Florida's streams and rivers and included here are the Suwanee, Apalachicola and other rivers which have extensive drainage areas. Wood and Fernald (1974) list twenty-eight major drainage basins and streams in Florida. The two largest, the Apalachicola River Basin and the Suwanee River Basin drain 50,777 sq km (19,600 sq mi) and 25,984 sq km (10,030 sq mi), respectively, some of which lies outside of Florida. Wood and Fernald (1974) indicate that peak annual flows occur in eastern rivers in September and October with buildups since June while more western rivers (Chipola, Apalachicola) reach maximum flow during March and April. In northwestern Florida a continental weather pattern predominates whereas a more

tropical weather routine predominates in central and south Florida.

Beck (1965) recognized five stream types in Florida: sand-bottomed, calcareous, swamp and bog, larger rivers, and canals. He points out that due to velocity, substrate, and spring discharges many of these types of flows can be recognized at different points along a single watercourse and cites the Suwanee River which is successively a swamp and bog stream, a sand-bottomed stream, a calcareous stream and a larger river of mixed origin. Beck (1965) classifies swift flow in Florida's streams as that velocity suitable for Plecoptera while simuliid populations indicate conditions of moderate velocity. Below these two levels the speed is considered low. A general progression is recognized with the swifter portions of upper rivers marked by eroding limestone and thick growths of moss, midpoints along rivers being sand-bottomed and lower, slower portions of rivers containing deposits of finer materials on the bottom. Beck (1965) lists the sand-bottomed stream as the most common type of stream in Florida. The fauna consists of Hydropsychid and Philopotamid caddisflies, simuliid larvae, Plecoptera, *Stenonema* mayflies, orthocladine chironomids, and *Corydalis*. In these sand-bottomed streams the pH reaction is usually between 5.7 and 7.4 and they display moderate to high color and moderate to swift velocity. The bottoms consist of sand, leaf deposits, and limestone outcroppings, and plant growth may be dense. Beck mentions that swamp and bog streams are highly acidic (pH reaction 3.8-6.5), sluggish flows found in the coastal lowlands and central highlands with origins in swamps and marshes. Calcareous streams are cool, clear flows of spring origin with dense growths of vegetation. These flows are alkaline (7.0-8.2 in pH), bear large mollusc populations, are generally of low color, of variable

velocity and have sand, clay or limestone bottoms. The flows in the larger river category usually carry considerable silt and are turbid, have high and steep banks and bottoms of coarse sand and limestone. They demonstrate a pH reaction of 6.5-7.4 and have few shallow places and few aquatic plants. Beck (1965) mentions from about the area of the St. Lucie Canal to south of Homestead no natural streams of any consequence remain along the east coast of Florida and, instead, canals are prevalent although in the past the Miami River had a swift flow and rapids (Fairchild, 1976 — personal communication).

The above discussion provides an introduction to the climatic, edaphic, biotic and hydrologic conditions which are found in the State of Florida.

Leucocytozoon smithi

During the 1880's Danilewsky observed parasites which were later described and designated as species of *Leucocytozoon* and *Haemoproteus* (Bennett et al., 1965). Hsu et al. (1973) list 67 species of *Leucocytozoon*, one of which is reported from *Meleagris gallopavo*, the domestic and wild turkey. Theobald Smith in 1895 first reported a protozoan-parasite in the blood of turkeys in Massachusetts and Rhode Island (Smith, 1895). Similar gametocytes were observed in the blood of turkeys in France and named *Haemamoeba smithi* (Laveran and Lucet, 1905). The following taxonomic scheme which shows the position of the turkey parasite now known as *Leucocytozoon smithi* was modified from Aikawa and Sterling (1974) by crediting Sambon (1908) not Danilewsky (according to Bennett et al., 1975) with the genus *Leucocytozoon*:

Phylum — Protozoa Goldfuss, 1818, emend. von Siebold, 1845
Subphylum — Sporozoa Leuckart, 1879
Class — Telosporea, Schaudinn, 1900
Subclass — Coccidia Leuckart, 1879
Order — Eucoccida Leger and Duboscq, 1910
Suborder — Haemosporina Danilewsky, 1885
Family — Leucocytozooidae Fallis and Bennett, 1961
Genus — *Leucocytozoon* Sambon, 1908
Species — *smithi* (Laveran and Lucet, 1905).

The following authors observed *L. smithi* in turkeys in their respective states or countries: Atchley (1951), Bierer (1954), Jones and Richey (1956) and Wehr (1962) in South Carolina; Savage and Isa (1945) and Bennett et al. (1965) in Canada; Volkmar (1929) in Minnesota and North Dakota; Hinshaw and McNeil (1943) in California; Johnson (1945) in Michigan; Kozicky (1948) in Pennsylvania; Skidmore (1931) in Nebraska; Stoddard et al. (1952) in Georgia; and Byrd (1959) in Virginia. Volkmar (1929) also lists *L. smithi* from Germany and the Crimean Peninsula and Cook (1971) lists Asia. Travis et al. (1939) reported *Leucocytozoon smithi* from wild and domestic turkeys in Florida as well as Georgia, Missouri, Alabama and South Carolina. Simpson et al. (1956) and Forrester et al. (1974) also reported *Leucocytozoon smithi* from Florida turkeys. Solis (1973) exposed turkeys, ringnecked pheasants, chickens, bobwhite quail, pekin ducks and chukar partridges in an area where *L. smithi* occurred and found that only turkeys were susceptible to *L. smithi*.

Fallis et al. (1974) and Aikawa and Sterling (1974) present information on the life cycle and structure of the intracellular parasite, *L. smithi*. *Leucocytozoon* belongs to the same suborder, Haemosporina, as *Plasmodium* and *Haemoproteus* and undergoes a similar life cycle. Unlike *Plasmodium*, however, there is no erythrocytic schizogony and in addition to the red blood cells being invaded by merozoites as in *Haemoproteus* (and *Plasmodium*), in *Leucocytozoon* white blood cells are

also used as sites for gametogony (Huff, 1942; Fallis et al., 1974). Gametocytes, in the case of *L. smithi*, grow and split the host nucleus in two (Sambon, 1908; Volkmar, 1929). *Plasmodium* species are transmitted by mosquitoes and *Haemoproteus (Parahaemoproteus)* species by the Hippoboscidae and Ceratopogonidae (Fallis and Bennett, 1961a and b; Bennett et al., 1974). *Leucocytozoon* is known at present to be transmitted only by black flies. A parasite of chickens which is vectored by *Culicoides* was designated *Akiba caulleryi* by Bennett et al. (1965) but *Akiba* has recently been considered as a subgenus of *Leucocytozoon* (Hsu et al., 1973; Fallis et al., 1974). Gametocytes are ingested with the blood of an infected turkey by a feeding black fly. Roller and Desser (1973) observed diurnal periodicity with *L. simondi* where peak gametocytemia corresponded with peak activity and biting periods of the vector. It has been suggested that there is periodicity of the gametocytes of *L. smithi* in the peripheral blood and correlation with the periodicity and feeding peaks of the vectors (Moore et al., 1974). Early in the infection each gametocyte appears as a round body in the parasitized cells; later the gametocytes deform each blood cell into the characteristic spindle shape (Huff, 1942). Desser et al. (1970) found with *L. simondi* elongated cells resulted when merozoites developed in leucocytes and round cells resulted from invasion of red blood cells. Macrogametocytes stain more darkly with Giemsa stain than microgametocytes. Within minutes of ingestion the microgametocytes and macrogametocytes escape from their host blood cells. The microgametocytes exflagellate and syngamy occurs with the macrogametes. The zygotes formed grow into elongate ookinetes within twelve hours after the initial blood ingestion. Sections of flies have revealed ookinetes in the process of penetrating

the midgut of the fly (Fallis et al., 1974). Fallis and Bennett (1961a) working with *L. simondi* detected only sluggish movement of ookinetes and expressed doubt that this motion could facilitate penetration of the gut wall of a black fly. Howard (1962) working with *Plasmodium gallinaceum* observed no mobility or active penetration by zygotes. Howard suggests a largely passive incorporation into the gut wall to within 5 microns of the external basement membrane as the blood meal is digested and the distended gut returns from a squamous cell configuration to its original columnar cell morphology. Desser (1970) described a pore, strengthening struts, microtubules and elongate convoluted micronemes in the apical cap of the anterior end of *L. simondi* ookinetes and suggested these structures might aid penetration through the peritrophic membrane and into the fly midgut epithelium. In as little as 48 hours the ookinetes develop into oocysts containing 50-100 sporozoites just beneath the outer membrane of the gut. Sporozoites escape from the oocysts and penetrate and collect in the salivary glands. Desser (1970) describes structures on the anterior end of sporozoites of *L. simondi* which may contain proteolytic enzymes and aid in entering the salivary glands and, later, host liver cells. The sporozoites pass out the proboscis of the black fly with the salivary fluids when the fly bites another host. The sporozoites of *L. smithi* invade the liver parenchymal cells of the turkey and grow into hepatic schizonts. Huff (1942) reports with other species of *Leucocytozoon* megaloschizonts in the heart, spleen and other host tissues. Such schizonts are not normally observed with *L. smithi*; however Siccardi et al. (1974) reported finding megaloschizonts in the kidneys of infected turkeys fed coccidiostat medication. The schizonts by asexual multiplication (schizogony) produce merozoites

which enter red and white blood cells. Peters (1971) suggests that in the related genus *Plasmodium* merozoite penetration may involve proteolytic enzymes and be active or may be more passive with the merozoites being engulfed or invaginated into the blood cells. The merozoites grow into gametocytes in the blood cells and the cycle is completed. The prepatent period or time elapsed between entrance of the sporozoites and appearance of gametocytes in the peripheral blood is 10 to 16 days.

Symptoms associated with leucocytozoonosis include: loss of appetite, emaciation, wheezing breathing, congested heart and lungs, drooling, drooping wings, sitting on the hocks, enlarged liver and spleen, anemia, impaired immunological responsiveness, and death (Wehr, 1962; Salsbury, 1971; Siccardi et al., 1974). Turkeys under 12 weeks of age are often severely affected but mortality in older birds has also been reported (Simpson et al., 1956). Many birds with high parasitemias appear outwardly normal. Birds that do not die may continue to carry the parasite for as long as 1½ years (Skidmore, 1931). Borg (1953), Simpson et al. (1956) and others suggest that the pathogenicity of *L. smithi* has not been proven conclusively and that *L. smithi* may be an additive debilitating factor which when combined with other factors such as blackhead, fowl cholera or leukosis results in bird mortality. Stoddard et al. (1952), Skidmore (1931), Savage and Isa (1945) and others however have observed flock mortality up to 75% where bacterial cultures and other tests revealed *L. smithi* as the sole disease organism present. Jones and Richey (1956) indicated annual death losses due to *Leucocytozoon* in one county in South Carolina averaged 5%. In 1973,132 million turkeys were raised in the U.S. and farmers grossed $934 million (Poultry Digest, April 1974; Agricultural Situation,

November 1974). In the absence of severe threats by disease this industry will continue to grow as the public's demand for poultry and its products like turkey rolls and t.v. dinners as an alternative to beef increases.

A number of black flies have been incriminated as possible vectors of *L. smithi*. Skidmore (1931) incriminated *S. occidentale (=meridionale)* as a vector of *L. smithi* in Nebraska by grinding up a number of flies that had fed on infected turkeys, injecting them in saline into clean birds, and 12 days later observing gametocytes of *L. smithi* in the turkeys' blood streams. Johnson et al. (1938) in Virginia exposed uninfected turkeys to the bites of *S. nigroparvum (=jenningsi)* and obtained *L. smithi* transmission. Underhill (1944) further substantiated this work but Byrd (1959) was unable to infect clean turkeys by macerating and injecting *S. jenningsi* that had fed on diseased birds. Wehr (1953) by injection and Jones and Richey (1956) by fly-bite incriminated *S. slossonae* as a vector of *L. smithi*. Byrd (1959) succeeded in infecting turkeys with *L. smithi* by grinding and injecting females of *Prosimulium hirtipes* that had fed on infected birds. I have seen no further follow up of this work and Fallis et al.(1974) do not list any member of the former *P. hirtipes* complex as a vector of *L. smithi*. Noblet et al. (1972) ground up in saline and injected infected females of *S. congareenarum* into turkeys and incriminated this species as a suitable or possible vector for *L. smithi*. Savage and Isa (1945) and Anthony and Richey (1958) have reported leucocytozoonosis in the supposed absence of simuliids although other biting flies such as *Culicoides*, *Stomoxys*, *Diachlorus* and mosquitoes were present.

Control of leucocytozoonosis can be achieved by locating turkey flocks at least 12.8 km (8 mi) from known breeding locations of potential

vectors and in areas free from possible wild turkey disease reservoirs. Control of black flies by large scale aerial treatment of streams with larvicides has been shown to markedly decrease the level of parasitemias of *L. smithi* in sentinel turkeys (Kissam et al., 1975). Fine mesh wire screen enclosures might be a valuable preventive technique with smaller flocks. Clopidol, a coccidiostat, at .025 and .0125% in feed is effective in reducing the number of *L. smithi* parasites in turkeys as indicated by blood smears (Siccardi et al., 1974).

Investigators of leucocytozoonosis in turkeys in Florida have studied the presence, consequence, and distribution of *L. smithi* in the State but have not proven specific vectors. Travis (1939) reported that nearly 100% of the wild turkeys in low areas of Georgia and Florida were infected with *L. smithi* and suggested that an aquatic breeding insect might be involved. Simpson et al. (1956) investigating outbreaks of *L. smithi* in turkey flocks near Palatka, Florida, reported finding *S. slossonae* and a species #58 (=*S. jonesi* - Stone and Snoddy, 1969) breeding near the turkey flocks and suggested that *S. slossonae* might be an important vector. Davis et al. (1957) reported *S. slossonae* as prevalent in Florida. Forrester et al. (1974) found a 72-75% incidence of *L. smithi* in 484 turkeys more than one month old sampled from 10 localities in Florida. These authors also noted that a drop in the infection rate of *L. smithi* observed in wild turkeys in the Sikes Fisheating Creek Wildlife Management Area and other areas of the State in 1971 corresponded with low rainfall and low stream conditions and theorized that reduced numbers of potential black fly vectors may have resulted in the reduced prevalence of *Leucocytozoon* in turkeys.

CHAPTER III
MATERIALS AND METHODS

Black Fly Survey

Primarily, efforts in this survey concentrated on collecting larvae and pupae from Florida's streams and rivers. In Alachua County ten streams were selected and in nearby counties an additional twenty flows were chosen which were fairly permanent and represented a diversity of ecological conditions. An attempt was made to secure samples from each of these streams at least once a month. Approximately the same location was revisited each time to obtain some consistency in stream and substrate conditions in order to judge seasonal replacement phenomena and normal population changes. Other sites in south, southwest and west Florida were chosen, generally along highways or country roads for accessibility, which were visited at least quarterly to obtain records at all seasons of the year.

At each location specimens were removed with forceps or placed still attached to small portions of grass, twigs and so on into four-dram lip vials with neoprene stoppers. Each vial contained 80% ethyl alcohol and a code number penciled on a small slip of paper. The immatures, collected in 30 to 45 minutes were deemed sufficient to indicate numbers and species present. Dark pupae, indicating an advanced state of development, were placed in four-dram screw cap vials on a strip of paper toweling moistened in the stream and were held alive in order to allow the adults to emerge. Imagos could be reared very successfully in this manner from

older and frequently, even younger, more pale pupae. The forceps used were secured to my wrist by a string to prevent their loss when my balance was upset by the current, when I was wading and saw an occasional water moccasin, and at other critical times. A 1.53 m long rake and a modified, extendible, golf ball retriever were used to obtain substrate samples and specimens where it was impossible to wade.

At each collection location stream dimensions, velocity, pH, temperature, color, and substrate were recorded on special data cards along with indications of the attachment substrates of the immatures and an estimate of the population size. Temperatures were obtained with a metal-cased pocket thermometer, range -1° to 49°C (30° to 120°F). Initial attempts were made to determine the pH of the streams using two portable, electronic pH meters. After obtaining erratic, inconsistent results in the field with the meters, pH papers (pHydrion papers - Micro Essential Labs, New York) were tried with more success. These pH papers come in ranges such as 3.5-5.5 and 6.0-8.0 between 1 and 12 pH units with illustrated color differences at .5 pH unit intervals and are rated accurate to .25 pH unit in buffered solutions. The papers were checked regularly for accuracy with pH 4.0, 6.5, 7.0 and 10.0 buffered solutions. Very acidic readings were confirmed by bringing chilled water samples into the lab and checking them on a Beckman pH meter. Experiments with electronic, propellered Gurley flow meters proved them too cumbersome to set up and difficult to use in the streams which were often shallow and clogged with submerged vegetation. The floating cork technique suggested by Dalmat (1955) and others was used. A weighted cork 3.8 cm in diameter which floated low in the water and was connected to a 3.05 m (10 ft) long string was timed with a stop watch to determine the stream velocity

at different points where larvae and pupae were located. Most immature black flies were encountered on trailing vegetation at or near the surface and thus the floating cork velocity figures should reasonably reflect immature habitat conditions. A ruler one meter long with centimeter markings was used to determine stream depths and widths. The widths of larger flows were estimated by pacing across them, when possible, or by pacing across a nearby bridge. Other omnipresent items on collecting trips included an insect net and a snake bite kit.

Alcohol specimens and pupal vials from each site were placed with the data card into cloth bags and transported to the lab where identifications were made with the aid of a Bausch and Lomb stereomicroscope. Larvae were usually identifiable without dissection (except unraveling of the respiratory histoblast); however representatives of each species were dissected and mandibles, respiratory histoblasts, cephalic apotome, gular notch, antennae and fans, and anal sclerite and hooks were prepared in cellosolve and mounted in balsam under individual small cover slips. Adults which emerged were allowed to harden for a few hours and were then killed and placed in alcohol, prepared and mounted in balsam on slides, or pinned on minutin nadeln inserted in white polyporous pieces on #3 insect pins.

In addition to immatures, some adult collecting was undertaken using a variety of traps. Black fly adults have been obtained in Florida by other individuals using light traps and Malaise (flight) traps. In this research three types of adult fly traps were used: the Manitoba trap (Thorsteinson et al., 1965), the blackout box trap (Anderson and Dicke, 1960), and a modified ramp-type trap. The latter two types of traps will be discussed with the *Leucocytozoon* work. A modified Manitoba

trap (Fig. 1) was constructed using 3 bamboo poles, each 2.1 m (7 ft) long, with a hole drilled vertically into the uppermost node of each pole into which a leg of a metal tripod laboratory ring stand was inserted. A triangular serex or organza canopy 1.2 m (4 ft) tall was positioned inside the leg frame and anchored at the three lower corners by strings tied through a hole in each pole. The apex of the tapering canopy with a 5.1 cm (2 in) wide elastic opening was drawn up a short distance through the ring opening and stretched around the lower lip of a 17.8 cm (7 in) tall lantern jar, the collecting vessel, which rested on the ring. The jar was topped with a double layer of white serex or organza held in place with rubber bands. The jar contained an inverted plastic funnel affixed to the jar with hot plastic glue. On strings attached around the base of the jar and hanging into the canopy was suspended a 1.2 m (4 ft) long cord with a black, cylindrical (20 cm wide, 22 cm tall) metal can or a black cardboard triangle, 63 cm on each side, on the end. A 23 cm wide black plastic "skirt" was sometimes fitted over the top of the canopy and positioned at the lower margin of the canopy to accentuate the "window" effect. A 31 cm long cloth bag which contained 1.36-1.8 kg of dry ice was also hung inside the canopy. The dry ice and the black target attracted Simuliidae and other Diptera which flew or walked up the canopy into the collecting jar. Adults could be aspirated from the jar by removing the outer cloth layer and inserting an aspirator tube through a small slit in the lower layer of material. Specimens could also be taken to the lab in the glass jar and immobilized by cold in a refrigerator, though condensation inside the jar often wet the specimens. Manitoba trapping was accomplished using one or two traps at a time in six counties at twelve locations for one

Figure 1. A modified Manitoba trap with a black plastic skirt.

to eight hour periods during the months of January through October. The air temperature, wind speed and relative humidity were measured during the collecting periods. Trapped adults were pinned or mounted on slides as with reared adults. Identification of immatures and adults was possible using taxonomic keys in Stone and Snoddy (1969), Stone (1964) and smaller publications on individual species.

Leucocytozoon smithi Transmission

In the investigation of the transmission of *L. smithi* to turkeys in Florida, modifications of Koch's postulates were used as guidelines. The first aim was to establish an association in nature between the host, the disease, and the vector. It was necessary to have a suspected vector, preferably a clean, reared one, feed on an infected host in the lab and to observe the stages of the parasite, especially the infective stage, the sporozoite, in the fly. A clean host was then to be infected by a vector which had become infected in the lab with the *Leucocytozoon* parasite being recovered in the gametocyte stage from the formerly uninfected turkey.

In order to confirm the reported presence of *L. smithi* in certain areas such as the Lochloosa Creek and Fisheating Creek Wildlife Management areas (L. Williams, 1974 — personal communication; Forrester et al., 1974), I set out sentinel turkeys. The same was done to establish the presence of the disease in other areas, and to obtain infected hosts to serve as donors for transmission studies. These turkeys, like all clean poults used in the transmission studies, were either hatched from fertile eggs in a fly-proof room or obtained as disease-free one-day old poults and held in a fly-proof room until use. Difficulties in obtaining clean

poults for experiments early in 1975 when winter and early spring species, suspected to be *L. smithi* vectors, were flying were overcome for work in early 1976. Usually three birds were placed out in the field near a black fly stream in a cage for about a week and provided with food and water (Fig. 2). There were three primary sentinel sites, all in Alachua Co., and additional birds were also set out for short periods at Fisheating Creek, Glades Co. (see Table 5). Sentinels were also placed out on a few occasions in a ramp trap described below. In addition I worked with Dr. D.F. Forrester at Fisheating Creek during 1974 when he exposed many young turkeys for two week periods in cages in cypress trees and on the ground and in 1975 when cages and ramp traps were used for turkey exposures.

To determine the species of black flies generally present at the sentinel sites, immature stages were collected and adults on the wing were sampled with Manitoba traps as described in the previous section. To identify black flies specifically attracted to turkeys I used blackout box traps (Fig. 3), ramp traps (Fig. 4) and exposed turkeys (Fig. 5). Anderson and DeFoliart (1961) used the technique of blackout box trapping in Wisconsin. In Florida the blackout box traps were used at two locations in 1974 and an additional seven sites during 1975 (see Table 7). A turkey was exposed in a cage on the ground or on a platform in a tree for about 15 minutes. A large cardboard box with two plastic collecting cups on the top which appeared as bright areas inside the otherwise dark box was then placed over the cage and bird. Black flies completing their blood meals or unfed but on the turkey exhibited a positive phototactic response, flew to the collecting cups and were captured. The flies were removed to holding cartons, the turkey was re-exposed, and the process was repeated.

Figure 2. Sentinel turkeys in an exposure cage.

Figure 3. A blackout box trap in the field.

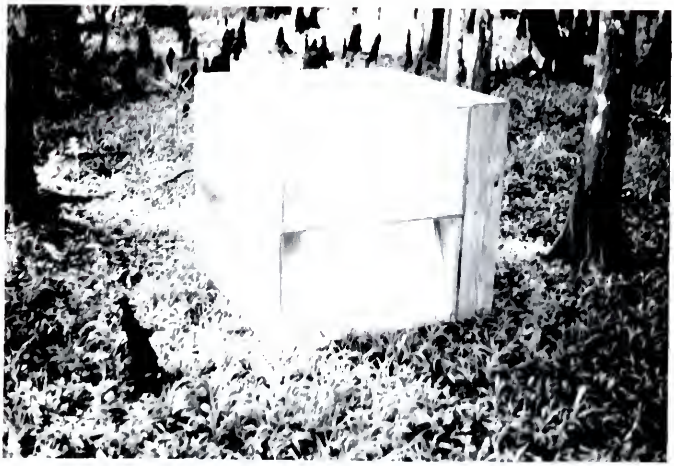

Figure 4. One view of a ramp trap.

Figure 5. An exposed turkey in the field.

An omnidirectional ramp trap was used with the hopes that black flies would be captured without the constant presence of the collector. The trap was used at three locations from April to July 1975 (see Table 6). The ramp trap was a wooden-framed cube approximately .92 m (3 ft) in each demension with 4 slanted lower level ingress panels of aluminum framing and organza material and 4 upper, vertical organza-paneled windows. A cage with host animals was placed and suspended inside the trap through a hinged access opening cut into the solid plywood top. The attracted insects would fly or bounce up the slanted ingress openings, enter the trap to feed on the hosts, theoretically be unable to escape and later be aspirated from the inside of the bright vertical cloth panels or windows.

By capturing flies off normally one or two exposed turkeys it was possible to tell which species approached the birds and, more significantly, to observe which species fed on the birds. Capturing flies off exposed turkeys first involved immobilizing the host. Masking tape was wrapped around the turkey's legs just above the feet which were thus held together. The bird was placed on its side and a layer of gauze followed by a strip of wide masking tape was placed across the turkey's neck, chest, and legs and affixed to the platform which was usually a large plastic tray. The upper wing was propped up to provide the black flies with easy access to bare skin. Using this technique it was possible to monitor the number and type of ornithophilic adults present and capture them as well as observe their feeding locations, duration and behavior. Their feeding could also be assisted and securing of blood-fed flies from exposed infected turkeys could be more readily ensured. To do this, flies that landed on the feathers of the wing or

elsewhere were quickly aspirated before they could scurry beneath the feathers and were placed in the glass aspirator tube against the bare skin of the underside of the wing elbow, the bare skin of the chest over the rib cage or against the turkey's neck and allowed to feed to repletion. The fly was observed, the feeding timed, and the turkey soothed and kept as still as possible to assure a complete blood meal. Once fed, the flies were transfered to paper pint ice cream cartons with netting tops, provided a moistened paper towel to maintain high humidity and transported to the lab. The exposed turkey technique was used to collect black flies on numerous occasions from January through August at 8 locations in 3 counties (Table 8).

To obtain black flies that had not been exposed to infections in the field and to obtain specimens of known identity, especially when working with species in the *Simulium* subgenus *Phosterodoros* which are difficult to distinguish as adults and are much easier to separate as pupae, adults were reared in the lab. Some adults were reared in pupal vials from field collected pupae and other adults were reared form field collected larvae and pupae in an aquarium with aerated water and a fine mesh netting cover.

Black flies which fed on exposed infected hosts in the field were held in the lab for at least two days after feeding and then allowed to bite an uninfected bird. All adult flies maintained in the lab were held in paper pint-sized ice cream cartons covered with netting and provided with moist cotton for water and a raisin for nutrition. Up to four cartons were kept at one time together in either a large plastic-covered aquarium or a glass desiccator-type container (Fig. 6) where high humidity was maintained by including a sponge half immersed in a

Figure 6. Glass container and paper cartons used for holding black fly adults alive in the laboratory.

cup of water. Flies captured in an unfed condition in the field or reared in the lab were allowed to feed on an infected turkey, held for two to seven days and then allowed to feed on a clean bird. All laboratory feedings involving uninfected hosts were conducted in a fly-proofed former bull room at the Veterinary Science area at the University of Florida. This room was incandescent lighted, had a double door entrance and had windows covered with very fine mesh organza material. Feedings on infected hosts were conducted either in the Veterinary Science room or at the fluorescent-lighted Veterinary Entomology Lab on campus. Black flies were routinely taken off their sugar source at least 24 hours before planned feeding trials and maintained only on the water provided by the moist cotton. Black flies were aspirated from the cartons or chilled in a refrigerator and removed with forceps and placed, usually one per vial, into seven-dram, clear plastic, cylindrical vials covered with green, fine mesh (30/6.45 sq cm) netting at both ends and held against the bare skin of the immobilized turkey. Room temperature, relative humidity and feeding times were recorded.

Uninfected turkeys bitten by possible vectors were held in a fly-proof room and the appearance of gametocytes in the peripheral blood was monitored by blood smears obtained by wing vein punctures every 3 to 4 days. Blood smears were processed by air drying, fixing in absolute methanol and staining in a 1:50 dilution of Giemsa solution and distilled water. Flies were dissected in .9% physiological saline and midguts and salivary glands were observed for the presence of oocysts and sporozoites of *L. smithi*. A few midguts were fixed in 10% formalin, stained in Haematoxylin and mounted in balsam, with the necessary intermediate steps for dehydration and clearing, in attempts

to more readily view the oocysts. Most midguts were air dried, fixed in methanol and stained with Giemsa, as were the salivary glands. The preparations were covered with permount and a cover slip for microscopic examination. Other fly structures were placed in 10% NaOH (except the wings) for 48 hours, rinsed in water and acid alcohol, placed in 100% cellosolve for 15 to 30 minutes and mounted in balsam on a microscope slide.

CHAPTER IV
RESULTS AND DISCUSSION

Simuliidae

General Comments

The primary efforts of this survey and ecological investigation of the black flies of Florida were concentrated on the immature stages. Over 700 adults were reared and pinned or mounted in balsam on slides and a number of adults have been captured in traps or on host animals (Tables 4, 6, 7 and 8) however the vast majority of specimens are alcohol - preserved larvae and pupae. Over 1,100 four-dram vials containing about 50,000 specimens from 1,080 individual, positive collections which have been examined and identified are on hand. In addition, records were obtained for Florida black flies from institution and individual collections and are included under the appropriate species.

Table 1 is a list of the Simuliidae collected during this research in Florida. Eighteen species representing two genera and six subgenera are recorded. Figure 7 shows the 192 locations where I personally collected specimens or obtained records from the collections of other individuals. One star normally designates a single collection site; in a few cases a single star is used to indicate two collection sites adjacent to each other such as a main stream and a side drainage flow to that main stream. Each collection location is listed by county and site number in the Appendix and the name and location of each stream is given. The 50 counties from which black flies are recorded are listed in Table 2

Table 1. Florida black fly species.

Cnephia (Cnephia) ornithophilia Davies, Peterson, and Wood*

Simulium (Byssodon) meridionale Riley

Simulium (Byssodon) slossonae Dyar and Shannon

Simulium (Eusimulium) congareenarum (Dyar and Shannon)

Simulium (Phosterodoros) dixiense Stone and Snoddy*

Simulium (Phosterodoros) haysi Stone and Snoddy*

Simulium (Phosterodoros) jenningsi Malloch

Simulium (Phosterodoros) jonesi Stone and Snoddy

Simulium (Phosterodoros) lakei Snoddy*

Simulium (Phosterodoros) notiale Stone and Snoddy*

Simulium (Phosterodoros) nyssa Stone and Snoddy

Simulium (Phosterodoros) taxodium Snoddy and Beshear*

Simulium (Psilozia) vittatum Zetterstedt*

Simulium (Simulium) decorum Walker

Simulium (Simulium) tuberosum (Lundström)

Simulium (Simulium) verecundum Stone and Jamnback*

Cnephia species undetermined No. 1*

Simulium species undetermined No. 1*

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*New collection record for Florida.

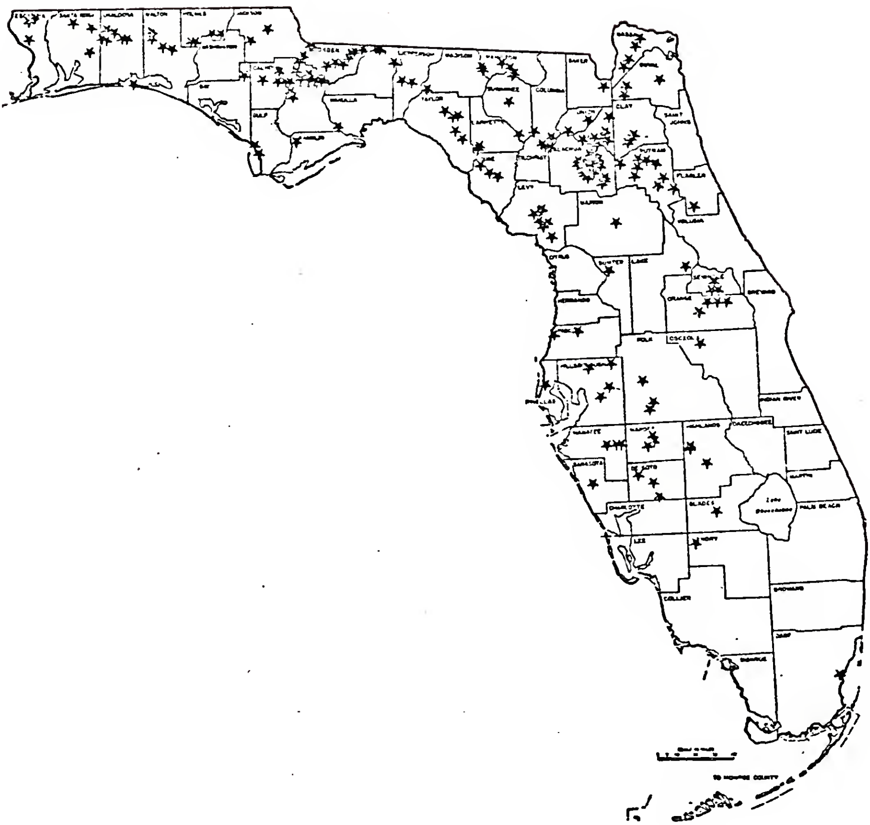

Figure 7. Locations in Florida where black flies have been collected.

Table 2. Florida black fly distribution records by county.

County	Sites	*C. ornithophilia*	*S. meridionale*	*S. slossonae*	*S. congareenarum*	*S. dixiense*	*S. haysi*	*S. jenningsi*	*S. jonesi*	*S. lakei*	*S. notiale*	*S. nyssa*	*S. taxodium*	*S. vittatum*	*S. decorum*	*S. tuberosum*	*S. verecundum*	*C. spp.* undetermined No. 1	*S. spp.* undetermined No. 1
Alachua	36	x		x	x			x	x	x			x	x	x	x	x	x	
Baker	1			x	x				x							x	x		
Bay	1			x					x							x			
Bradford	7	x		x	x				x	x			x	x		x	x		
Calhoun	5		x	x		x			x				x			x			
Clay	3			x					x							x			
Columbia	5			x				x		x			x			x			
Dade	1			x				x											
Desoto	3							x		x			x						
Dixie	3	x		x				x		x			x	x			x		
Duval	3	x	x	x	x				x				x	x		x	x		
Escambia	3			x		x			x							x	x		x
Flagler	1			x				x		x			x						
Franklin	1		x	x	x														
Gadsden	10			x					x		x			x	x	x	x		
Glades	1			x				x		x			x						
Gulf	2			x															
Hamilton	3	x		x	x			x		x			x	x		x	x		
Hardee	3									x			x	x		x			
Hendry	1							x		x									
Highlands	3			x												x			
Hillsborough	4			x				x		x			x	x					
Holmes	4	x		x	x				x			x	x	x		x	x		
Jackson	2		x	x															
Jefferson	2			x					x	x					x	x	x		
Lafayette	2			x	x			x		x			x	x			x		
Lake	1			x				x		x			x						
Leon	3	x		x	x									x	x	x	x		
Levy	7	x		x				x		x			x	x			x		
Liberty	10	x	x	x	x				x							x	x		
Madison	3			x	x			x		x			x	x		x	x		
Manatee	3			x				x		x			x			x			
Marion	1															x			

Table 2. (Continued)

County	Sites	*C. ornithophilia*	*S. meridionale*	*S. slossonae*	*S. congareenarum*	*S. dixiense*	*S. haysi*	*S. jenningsi*	*S. jonesi*	*S. lakei*	*S. notiale*	*S. nyssa*	*S. taxodium*	*S. vittatum*	*S. decorum*	*S. tuberosum*	*S. verecundum*	*C.* spp. undetermined No. 1	*S.* spp. undetermined No. 1
Nassau	4	x		x	x				x					x		x	x		
Okaloosa	7	x		x	x	x			x					x	x	x			
Orange	4			x								x				x			
Osceola	1			x															
Pasco	2			x										x					
Pinellas	1			x															
Polk	3			x				x		x			x	x		x			
Putnam	9			x				x	x	x				x		x	x		
Santa Rosa	3			x	x	x	x		x					x		x	x		
Sarasota	1			x															
Seminole	3	x		x				x		x			x	x		x	x		
Sumter	1			x						x									
Suwanee	3			x				x		x			x			x			
Taylor	5	x		x	x			x	x	x			x	x			x		
Union	2	x		x	x			x	x	x			x			x	x		
Wakulla	1			x															
Walton	4			x					x							x	x		

Table 3. Black fly associations based on collections of immature stages.*

	C. ornithophilia	*S. meridionale*	*S. slossonae*	*S. congareenarum*	*S. dixiense*	*S. haysi*	*S. jenningsi*	*S. jonesi*	*S. lakei*	*S. notiale*	*S. nyssa*	*S. taxodium*	*S. vittatum*	*S. decorum*	*S. tuberosum*	*S. verecundum*	*S. spp.* undetermined No. 1
C. ornithophilia			32	26			1	4	4			2	2		32	10	
S. meridionale								1									
S. slossonae	32			84	3	1	38	107	73			52	15		186	55	
S. congareenarum	26		84				2	19	7			6	2		49	23	
S. dixiense			3			3		24							21	4	
S. haysi					3			5							5		
S. jenningsi	1		38	2				19	92			73	4		21	8	
S. jonesi	4	1	107	19	24	5	19		36	2	1	40	11	3	253	44	
S. lakei	4		73	7			92	36				150	15		54	19	
S. notiale								2						2	3	1	
S. nyssa								1							1		
S. taxodium	2		52	6			73	40	150				8		44	10	
S. vittatum	2		15	2			4	11	15			8		9	50	33	
S. decorum								3		2			9		6	7	
S. tuberosum	32		186	49	21	5	21	253	54	3	1	44	50	6		78	1
S. verecundum	10		55	23	4		8	44	19	1		10	33	7	78		
S. spp. undetermined No. 1															1		

*By reading across or down from a selected species the number of times the immatures of that species were collected together with the immatures of another species is indicated by the figure at the intersection of the two columns.

Table 4. Black flies captured in Manitoba traps.

Date	Location	Collection times	Species (all females)
1974: 3 May	Univ. of Florida, Site 34	1730-2030	*S. vittatum*
30 May	Hatchet Creek Preserve, Site 24	1130-1930	*S. slossonae*, *S. (Phosterodoros) spp.*
28 June	Lochloosa Creek, Site 22	1800-2000	*S. slossonae*
28 June	Hatchet Creek Preserve, Site 24	1710-2020	*S. slossonae*
6 July	Junction SR 225/ 340, Site 8	1500-1745	*S. slossonae*
11-12 July	Fisheating Creek, Site 95	0630-0930; 1730-2045	*S. slossonae*, *S. (Phosterodoros) spp.*
16 July	Lochloosa Creek, Site 21	1630-1845	*S. slossonae*, *S. (Phosterodoros) spp.*
16 July	Hatchet Creek Preserve, Site 24	1600-1800	*S. slossonae*, *S. (Phosterodoros) spp.*
19 July	Sandy Hatchet Creek, Site 1	1600-1730	*S. slossonae*
19 July	Univ. of Florida, Site 34	1500-1830	*S. slossonae*
24 July	Hatchet Creek Preserve, Site 24	1530-1815	*S. slossonae*
26-27 July	Fisheating Creek, Site 95	0700-1010; 1530-1800	*S. slossonae*, *S. (Phosterodoros) spp.*
15 Aug	Lochloosa Creek, Site 22	1730-1845	*S. slossonae*, *S. (Phosterodoros) spp.*
17 Aug	Univ. of Florida, Site 34	0830-1430	*S. slossonae*
15 Sep	Lochloosa Creek, Site 22	1630-1805	*S. slossonae*, *S. (Phosterodoros) spp.*
1975: 24 Jan	Lochloosa Creek, Site 22	1445-1710	*S. congareenarum*, *S. slossonae*, *S. (Phosterodoros) spp.*

Table 4. (Continued)

Date	Location	Collection times	Species (all females)
1975: 26 Jan	Double Run Creek, Site 43	1000-1330	*S. congareenarum*, *S. slossonae*
12 Mar	Thomas Creek, Site 159	1100-1330	*S. congareenarum*, *S. slossonae*
12 Mar	8 km west of Highway 115C on Rt 90, Site 72	1000-1400	*S. slossonae*
6 May	Double Run Creek, Site 43	0930-1330	*S. slossonae*
6 May	Turkey Creek, Site 216	1030-1300	*S. slossonae*, *S. (Phosterodoros) spp.*
1 June	Junction SR 225/340, Site 8	0930-1200	*S. slossonae*
18 June	Hatchet Creek Preserve, Site 24	1400-1930	*S. slossonae*, *S. (Phosterodoros) spp.*
27-29 June	Fisheating Creek, Site 95	0730-1145; 1600-1910	*S. slossonae*, *S. (Phosterodoros) spp.*
7 July	Lochloosa Creek, Site 22	1730-2000	*S. slossonae*
29-30 July	Fisheating Creek, Site 95	0730-1200	*S. slossonae*, *S. (Phosterodoros) spp.*
9-10 Aug	Fisheating Creek, Site 95	0700-0830; 1500-1900	*S. slossonae*, *S. (Phosterodoros) spp.*
17-18 Sep	Fisheating Creek, Site 95	0830-1130; 1515-1715	*S. slossonae*, *S. (Phosterodoros) spp.*
15-16 Oct	Fisheating Creek, Site 95	0815-1315; 1620-2000	*S. slossonae*
1976: 17 Feb	Turkey Creek, Site 216	1625-1800	*S. congareenarum*, *S. slossonae*
24 Mar	Double Run Creek, Site 43	1400-1530	*S. congareenarum*, *S. slossonae*

Table 4. (Continued)

Date	Location	Collection times	Species (all females)
1976: 24 Mar	Turkey Creek, Site 216	1640-1830	*S. congareenarum*, *S. slossonae*
26 Mar	Sante Fe College, Site 28	1030-1800	*S. tuberosum*, *S. slossonae*

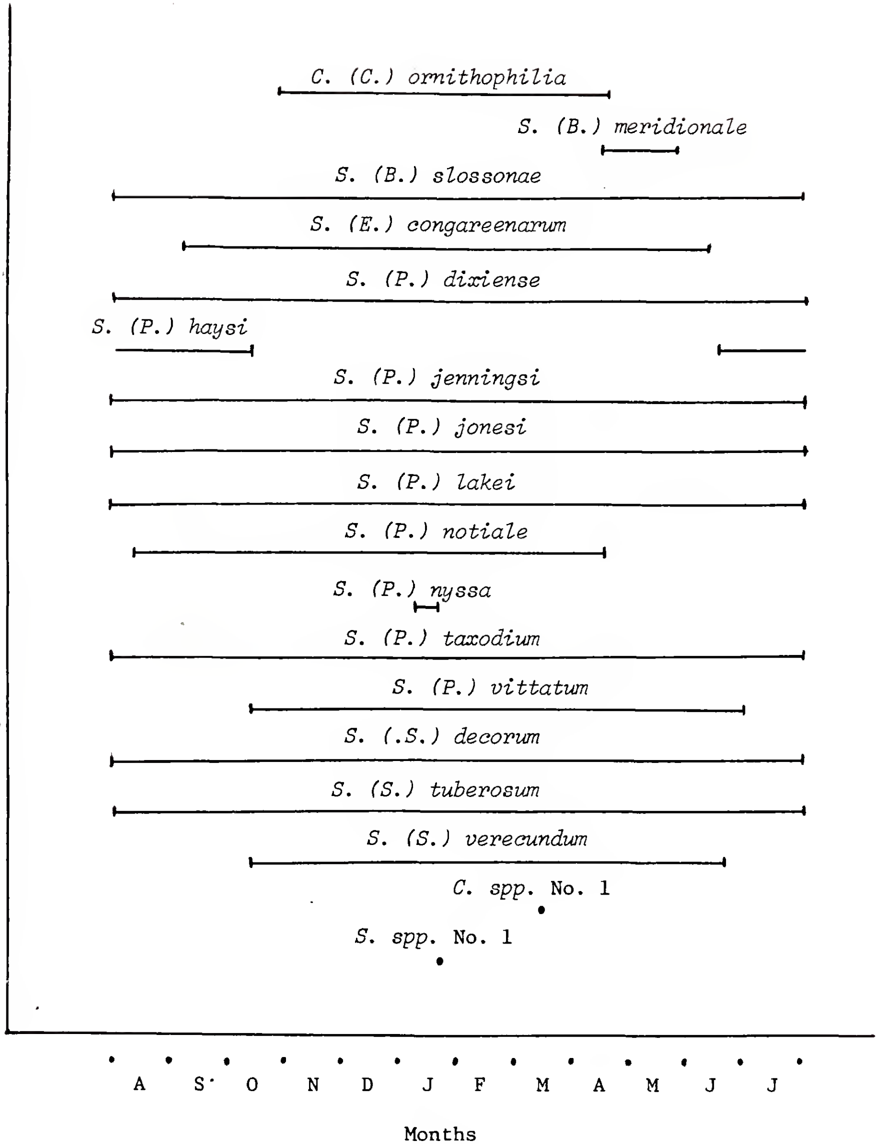

Figure 8. Seasonal occurrence of black flies in Florida.

with an indication of the species found in each county. Table 3 presents information on the frequency of association of black flies in Florida's streams and rivers. Figure 8 shows the seasonal occurrence patterns of the collected Simuliidae. Table 4 presents information on species captured in Manitoba traps. Further reference to these figures and tables will be made in discussing the individual species below. Keys to the black flies of Florida, in all stages, are included.

Introduction to the Black Fly Keys

Characters which are important taxonomically on black fly larvae and are noted on Figures 9, 10, and 11 include: the anterior, multiple-rayed cephalic fans and their stalks (CF,CFS); the antennae (A); the central dorsal section of the larval head capsule referred to as the frontoclypeus or the cephalic apotome (CA) which bears usually dark pigmented areas called head spots (HS); the ventral, setae bearing and anteriorly toothed hypostomium or submentum (S); posterior to the submentum is the throat (or postgenal) cleft (TC) or gular notch which differs in size and shape in the different species; laterally on the thoracic region are a pair of organs called the respiratory histoblasts (RH) consisting of a number of coiled filaments whose number and branching pattern are often diagnostic; the ventral proleg (VP); dorsally at the posterior end the anal gills (AG), osmoregulatory organs, protrude and may be simple and branchless stalks or may have many branches and be arborescent; behind the anal gills is located the anal sclerite (AS) or X-piece which assists the larva in releasing its posterior from the silk attachment patches; the circlet or many rows of tiny anal hooks (AH) at

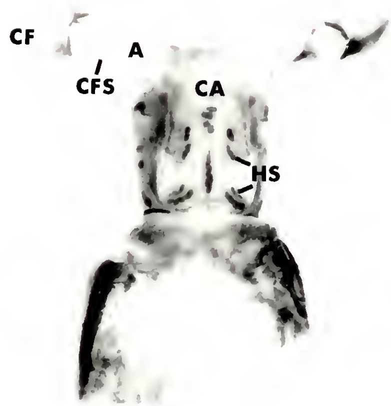

Figure 9. Dorsum of the head capsule of a black fly larva *(S. slossonae)*.

Figure 10. Venter of the head capsule of a black fly larva *(C. ornithophilia)*.

Figure 11. Lateral view of two black fly larvae *(S. dixiense)*.

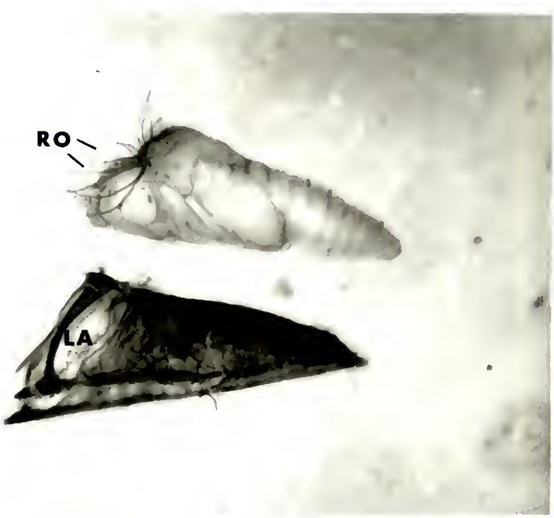

Figure 12. Black fly pupa and cocoon *(S. dixiense)*.

the very end of the larva; and the ventral protrusions which occur on some larvae and are called ventral or anal tubercles (VT).

Characters important on the pupa include: the filamented repiratory organs (RO); tiny setae-like structures on the thorax called trichomes ; and posterior dorsal tail hooks (Fig. 12). On the cocoon lateral apertures (LA) or windows occur anteriorly in the *Phosterodcros* species and the texture and general regularity of the cocoon is sometimes useful in separating species.

On the adult wing the presence or absence of hairs under the subcostal vein (SC); hairs on the dorsal, basal portion of the radius (R); the presence or absence of a basal cell (BC); and color of hairs on the stem vein (SV) are valuable characters (Fig. 13).

On the adult female head the shininess or pollinosity of the frons (F) and its shape and that of the clypeus (C), the color of the antennal segments (A), and the shape or size of the sensory vescicle of the maxillary palps (SV) are useful taxonomically (Fig. 14).

Important characters of the male genitalia include: the appearance of the ventral plate (VP) and the shape of its median portion; the shape and relative lengths of the basimere (B) and distimere or clasper (D); the presence or absence of a basal lobe on the distimere; and the number of distal spines (DS) or teeth on the distimere (Fig. 15).

On the female hind leg the presence or absence of a dorsal groove called the pedisulcus (P) on the second tarsal segment and the size of the calcipala (CL), a flattened lobe on the inner side and at the apex of the basitarsal segment, as well as presence or absence of a basal tooth (BT) on the tarsal claws are important characters (Fig. 16).

Structures useful for determinations in the region of the female

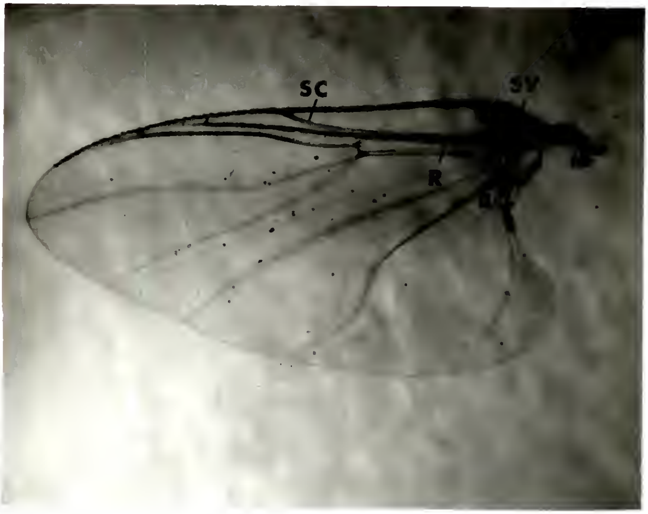

Figure 13. A wing of the black fly *Cnephia ornithophilia*.

Figure 14. A frontal view of the head of a female black fly (*S. notiale*).

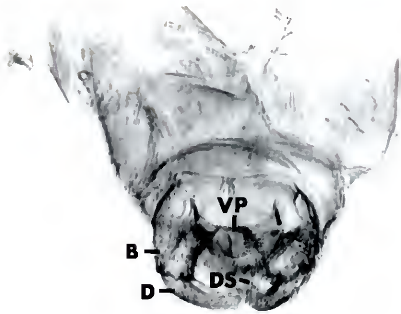

Figure 15. The male genitalia of a black fly, *Cnephia ornithophilia.*

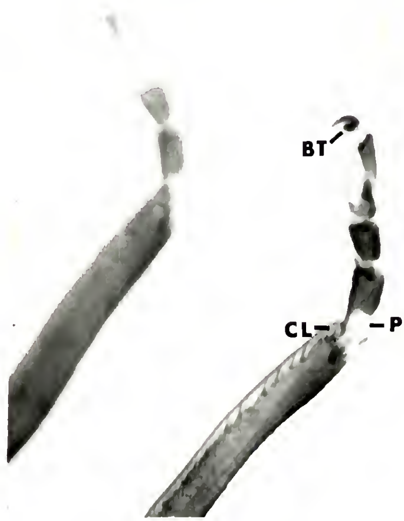

Figure 16. The distal portion of the hind leg of a *S. meridionale* female.

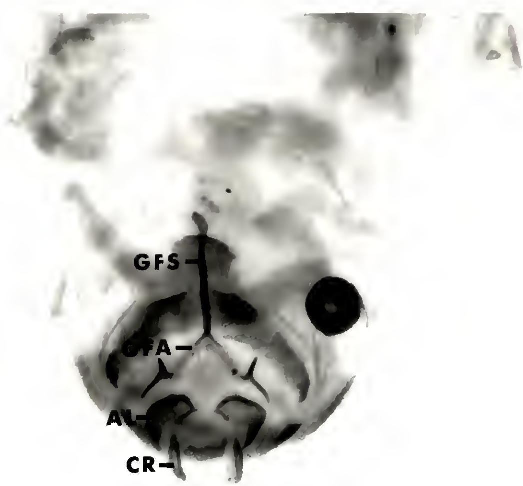

Figure 17. The terminalia of a female *S. meridionale*.

genitalia (Fig. 17) are the size and shape of the genital fork stem (GFS) and arms (GFA), the ovipositor lobes, each anal lobe (AL) and each cercus (CR).

A key to the larvae of the black flies of Florida.

(Photographic illustrations of certain structures referred to in this key and the keys which follow are included in the sections on the individual species. The larval key is primarily of value for later instars. For further illustrations refer to Stone and Jamnback (1955), Davies et al. (1962), Wood et al. (1963), Stone (1964), Stone and Snoddy (1969), Snoddy and Beshear (1968) and Snoddy (1971, 1976).

1a. Hypostomium convex along anterior margin; head spot pattern as in Fig. 18. . . . . *C. ornithophilia*

1b. Hypostomium concave or level along the anterior margin; the head spot pattern not as in Fig. 18 2

2a. (1b) Head spots light (white) 3

2b. Head spots dark or indistinct in a fulvous pattern 5

3a. (2a) Head spots consisting of a central, posterior white spot with dark rays projecting and diverging anteriorly (Fig. 247, Stone and Snoddy, 1969; no mature larvae collected). . . . . . .*S. meridionale*

3b. Head spots with anterior and posterior medial and lateral spots all white, spots visible in the usual positions. 4

4a. (3b) Medial anterior and posterior white head spots bordered by a dark fulvous area on each side (Fig. 99); respiratory histoblast with 8 filaments . . . . *S. decorum*

4b. No distinct dark fulvous border by central head spots; histoblast with 6 filaments . . . . *S. verecundum*

5a. (2b) Gular notch in the form of a shallow inverted -v; headspots as in Fig. 121 . . . . *C. species* No. 1

5b. Gular notch rectangular, sub-rectangular or sagittiform. 6

6a. (5b) Gular notch broadly sagittiform, extending at least across ½ of the venter of the head capsule, as wide as long 7

6b. Gular notch not sagittiform or else longer than wide 13

7a. (6a) Respiratory histoblasts consisting of 10 filaments 9 of which arise from a thick basal 10th (Fig. 60); anterior medial head spots often weak . . . . *S. jonesi*

7b. Respiratory histoblast not as above; all head spots usually dark, distinct 8

8a. (7b) Respiratory histoblast with 6 filaments; anal tubercles not prominent. . . . . *S. notiale*

8b. Respiratory histoblast with more than 6 filaments; anal tubercles prominent. 9

9a. (8b) Respiratory histoblast with 7 filaments. . . . . *S. haysi*

9b. Respiratory histoblast with more than 7 filaments. 10

10a. (9b) Respiratory histoblast with 8 filaments. . . . . . *S. taxodium*

10b. Respiratory histoblast with more than 8 filaments. 11

11a. (10b) Respiratory histoblast with 9 filaments . . . . *S. lakei*

11b. Respiratory histoblast with 10 filaments 12

12a. (11b) 10 respiratory histoblast filaments with a pattern as in Fig. 81; anal tubercles prominent and conical. . . . . *S. nyssa*

12b. 10 respiratory histoblast filaments with a pattern as in Fig. 53; anal tubercles small and rounded . . . . *S. jenningsi*

13a. (6b) Gular notch long extending ½ the distance or more to the teeth of the submentum, sagittiform or subrectangular, pointed or rounded anteriorly. 14

13b. Gular notch extending less than ½ the distance to the teeth of submentum, subrectangular. 16

14a. (13a) Head spots indistinct in fulvous area; gular notch usually with parallel sides; anal tubercles inconspicuous . . *S. tuberosum*

14b. Head spots distinct; gular notch with curved margins, elongate sagittiform; anal tubercles prominent 15

15a. (14b) Respiratory histoblast with 6 filaments; larvae reddish. . . . . *S. slossonae*

15b. Respiratory histoblast with 10 filaments; larvae yellow. . . . . *S. dixiense*

16a. (13b) Respiratory histoblast filaments 4 in number; gular notch longer than wide. . . . . *S. species* No. 1

16b. Respiratory histoblast filaments number more than 4; the gular notch not longer than wide. 17

17a. (16b) Respiratory histoblast with 12 filaments; the anal tubercles are large, prominent; a medium-sized larva, 6 mm long. . . . . *S. congareenarum*

17b. Respiratory histoblast with 16 filaments; the anal tubercles are absent or inconspicuous; a large larva, 8-9 mm long . . *S. vittatum*

A key to the pupae of the black flies of Florida.

(No pupae of *Cnephia species* No. 1 have been observed.)

1a. Cocoon a loose mass of silk, indistinct shape; strong dorsal hooks at the posterior end of the pupa. . . . . *C. ornithophilia*

1b. Cocoon a distinct slipper or pocket shape, strong dorsal hooks absent. 2

2a. (1b) Cocoon with large anterior, lateral apertures 3

2b. Cocoon uniform without large lateral apertures. 10

3a. (2a) Pupa with 6 respiratory filaments on each side; cocoon antero-ventrally completely joined (Fig. 75). . . . . . . *S. notiale*

3b. Pupa with more than 6 filaments 4

4a. (3b) Pupa with 6 filaments rising off a strong basal 7th one (Fig. 48) . . . . *S. haysi*

4b. Pupa with more than 7 filaments 5

5a. (4b) Pupa with 8 filaments with four pairs of 2. . . . *S. taxodium*

5b. Pupa with more than 8 filaments 6

6a. (5b) Pupa with 9 filaments in the pattern 2, 2, 2 and 3 from the dorsal. . . . . *S. lakei*

6b. Pupa with 10 filaments. 7

7a. (6b) Pupa with 9 filaments rising from a thick basal 10th one (Fig. 60) . . . . *S. jonesi*

7b. Pupal respiratory organ lacking a strong basal filament 8

8a. (7b) Pupa with 10 respiratory filaments which rise as 5 pairs, the petiole for filaments 7 and 8 (from the dorsal) noticiably longer

and stouter than the other petioles (Fig. 12) . . . . . *S. dixiense*

8b. Pupa with 10 filaments, some of which rise in pairs others in triplets. 9

9a. (8b) The lower more ventral respiratory filaments on long petioles, filaments long. . . . . *S. nyssa*

9b. Lower filaments rise close to the base, petioles short, filaments short . . . . *S. jenningsi*

10a. (2b) Pupa with 4 respiratory filaments. . . . . . . *S. species* No. 1

10b. Pupa with more than 4 filaments 11

11a. (10b) Pupae with 6 filaments in the respiratory organ. 12

11b. Pupae with more than 6 filaments. 14

12a. (11a) Pupal cocoon with concave anterior edge (in lateral view); respiratory filaments rising from long petioles (Fig. 28) *S. slossonae*

12b. Pupal cocoon with convex or straight anterior edges; petioles short 13

13a. (12b) Respiratory filaments widespread, base of filaments 5 and 6 sharply separated from base of 3 and 4, filaments long (Fig. 115). . . . . *S. verecundum*

13b. Respiratory filaments not widespread, bases about equidistant, filaments short (Fig. 108). . . . . *S. tuberosum*

14a. (11b) Pupa with 8 respiratory filaments; cocoon with rough texture. . . . . *S. decorum*

14b. More than 8 filaments 15

15a. (14b) Pupa with 12 respiratory filaments; cocoon with long anterior

dorsal projection (Fig. 35). . . . .*S. congareenarum*

15b. Pupa with more than 12 filaments; cocoon lacking dorsal projection 16

16a. (15b) Pupa with 16 filaments in 8 pairs . . . . *S. vittatum*

16b. Pupa with more than 20 filaments in pairs and in 3's .*S. meridionale*

<u>A key to the adult male black flies of Florida.</u>

(Adapted and modified from Stone and Snoddy, 1969)

1a. Second tarsal segment of the hind leg without a pedisulcus but a shallow depression may be present; radius vein with hair dorsally on the basal segment; large basal cell present . . .*C. ornithophilia*

1b. Second tarsal segment of the hind leg with a deep pedisulcus; base of radius with or without hair dorsally; large basal cell absent. . . . . *(Simulium)* . . .2

2a. (1b) Basal portion of radius with hair dorsally . *S. congareenarum*

2b. Basal portion of radius without hair dorsally. 3

3a. (2b) Distimere short, stout, with 3 or more teeth . . . *S. vittatum*

3b. Distimere long, with 1 tooth or none 4

4a. (3b) Distimere with a rounded lobe on the inner margin near the base 5

4b. Distimere without a lobe or basal projection 6

5a. (4a) Distimere with basal lobe bearing a number of stout, spine-like setae . . . . *S. tuberosum*

5b. Distimere with basal lobe bearing fine hairs only. . . *S. slossonae*

6a. (4b) Ventral plate in ventral view broadly rounded. *S. meridionale*

6b. Ventral plate in ventral view more narrow, compressed from the sides. 7

7a. (6b) Ventral plate with basal arms bearing distinct lateral projections; posterior third of scutum shiny with indistinct hairs. 8

7b. Ventral plate with basal arms without distinct lateral projections; posterior quarter or less of scutum shiny with some strong erect hairs. 11

8a. (7a) Ventral plate in ventral view with median portion longer than wide and parallel-sided or nearly so; scutum with silver spots narrow, oblique; the dark area between the spots broadens anteriorly. . . . . *S. dixiense*

S. jenningsi

S. notiale

8b. Ventral plate in ventral view with median portion not longer than wide and widened toward the end; anterior silver spots and intervening dark area variable. 9

9a. (8b) Scutum with large anterior silvery spots each extending about one-third the distance across the front of the scutum; the dark area between the spots narrows strongly to the anterior margin. .*S. jonesi*

S. taxodium

9b. Scutum with smaller silvery spots; the dark area between the spots is broad, converges little, and is narrowest about midway along the length of the spots . . . . *S. haysi*

S. lakei

S. nyssa

10a. (7b) Ventral plate in ventral view narrow, V-shaped, middle region tapers almost to a point; with a ventral keel. . *S. decorum*

10b. Ventral plate broader, not as strongly compressed in the middle section; ventral keel absent. . . . . *S. verecundum*

<u>A key to the adult female black flies of Florida</u>.

(Adapted and modified from Stone and Snoddy, 1969)

1a. The second hind tarsal segment lacks a pedisulcus, although there may be a slight depression; the basal cell is present and the basal portion of the radius bears setae dorsally . . . . *C. ornithophilia*

1b. The second hind tarsal segment with a deep pedisulcus; large basal cell absent and the basal portion of the radius bears or lacks setae dorsally (*Simulium*). 2

2a. (1b) Radius vein with hair dorsally on the basal section. . . . . *S. congareenarum*

2b. Radius without hair dorsally on the basal section. 3

3a. (2b) Tarsal claw with a prominent basal tooth 4

3b. Tarsal claw simple, without a prominent basal tooth. 5

4a. (3a) Frons gray pollinose . . . . *S. meridionale*

4b. Frons shiny black. . . . .*S. slossonae*

5a. (3b) Frons and terminal abdominal tergites shiny black or dark brown. 6

5b. Frons and terminal abdominal tergites at least lightly pollinose.10

6a. (5a) Subcostal vein with a row of hairs ventrally; scutum subshiny 7

6b. Subcostal vein without hairs ventrally; scutum shiny 8

7a. (6a) Fore tibia with a narrow gray-white patch not more than one third the width of the tibia; inner margins of ovipositor lobes fairly straight. . . . . *S. tuberosum*

7b. Fore tibia with a brilliant white patch which covers one half or more of the tibia; inner margins of the ovipositor lobes concave enclosing an oval area . . . . *S. verecundum*

8a. (6b) In anterior view the scutum displays a pair of fairly distinct rounded pollinose spots with a darker area of the scutum in between; hairs on the stem vein are dark brown to black . . . . *S. jenningsi*

S. notiale

S. nyssa

S. taxodium

8b. In anterior view the scutum is not pollinose or the pollinosity occurs as a diffuse area along the front and sides 9

9a. (8b) Clypeus about as wide as long. . . . . *S. jonesi*

S. lakei

9b. Clypeus longer than wide . . . . *S. dixiense*

S. haysi

10a. (5b) Scutum silvery gray with dark brown markings; abdomen with a black and light gray pattern . . . . *S. vittatum*

10b. Scutum uniform brownish gray without contrasting dark brown markings; abdomen black with thin gray pollinosity without a pattern. . . . . *S. decorum*

Introduction to the Individual Species Sections

The following sections deal specifically with the black fly species found in Florida. References are listed below each species name which provide synonyms for the currently accepted name and additional sources of descriptions and figures. The descriptions which follow the brief taxonomy portions are intended to point out outstanding or diagnostic characters and are accompanied with photographic illustrations. Under "Florida Observations" below the heading "Stream Parameters" a summary is given of the dimension, pH, temperature, and velocity figures for streams which contained each particular species. In the section on Florida collection records the numbers immediately following the county names refer to individual collection sites identified in the Appendix. The code for the collection records is as follows: S refers to small, very young larvae with no or weakly developed respiratory histoblasts; M refers to medium-aged larvae with distinct but white histoblasts; L refers to large, mature larvae with dark histoblasts; P refers to full pupae or pupal exuviae; C refers to cocoons; and A refers to male or female adults. The majority of records refer to specimens the author personally collected; in those instances where specimens were gathered by other individuals, the collector is noted. On the individual species maps when a record is included with the only locality information being the county the mark for that record on the map is placed in the center of the county. Immature, very young larvae lacking well-developed respiratory histoblasts in the subgenus *Phosterodoros* are difficult if not impossible to differentiate as are the adult females. Records for the *Phosterodoros* species are based primarily on mature larvae and pupae. Only at locations where

essentially a single *Phosterodoros* species was found to exist are records for earlier stages included. Records for undetermined species are listed as *S. (Phosterodoros) spp.* in the Manitoba trap results (Table 4). Dr. E.L. Snoddy kindly examined immature and adult specimens of *S. jenningsi*, *S. lakei* and *S. taxodium* from Florida and confirmed my identifications.

Cnephia (Cnephia) ornithophilia Davies, Peterson, and Wood

Cnephia ornithophilia Davies, Peterson and Wood, 1962, Proc. Entomol. Soc. Ontario 92: 102 (female).

Cnephia ornithophilia — Stone and Snoddy, 1969, Auburn Univ. Agr. Exp. Sta. Bull. 390: 25 (female, larva).

Taxonomy. Davies et al. (1962) first described a female of this species. The holotype was collected from a blue jay at Algonquin Park, Ontario and was deposited in the Canadian National Museum. Paratypes are located in the U.S. National Museum. Stone and Snoddy (1969) indicated the male of *C. ornithophilia* was not known and that the larva and pupa were apparently indistinguishable from those of *C. pecuarum*.

Description. The larva is large, about 8 mm long with a grayish brown abdomen and a light yellow brown head capsule. The cephalic spots are dark with the median groups of about 20 forming one long continuous row (Fig. 18). The submentum is convex apically and bears small teeth. The gular notch has fairly parallel sides, is broadly rounded anteriorly and extends less than 1/3 the distance to the submental teeth (Fig. 10). The cephalic fans each contain about 60 rays with long hair-like spines. The anal tubercles are absent.

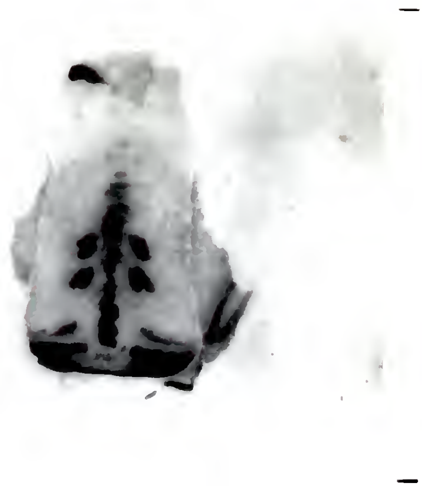

Figure 18. The head spots of a larva of *C. ornithophilia.*

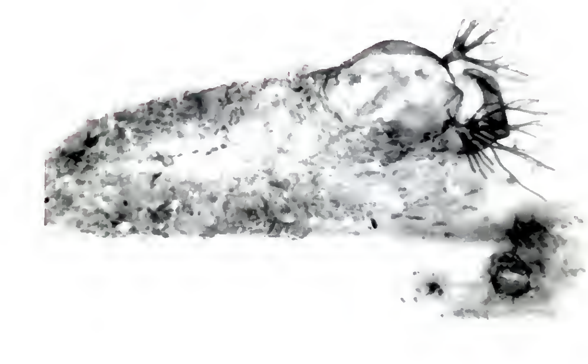

Figure 19. The pupal exuvium and cocoon of *C. ornithophilia.*

The pupa bears strong dorsal posterior hooks and is located in an irregular, loosely woven cocoon. The respiratory organs each contain about 30 filaments (Fig. 19).

This is believed to be the first description of the male. The wings are 3.75-4 mm long. There are hairs dorsally on the base of the radius. The first segment of the flagellum is the longest, about twice the length of the other segments. The scutum is dull brownish black with numerous short golden hairs. The scutellum is reddish brown and bears long dark hairs. The legs are almost uniform yellow brown. The second hind tarsal segment lacks a pedisulcus. The abdomen is dull brownish black dorsally and lighter gray or light brown ventrally. The ventral plate in ventral view is broad and the tapering distimere which is almost equal in length to the basimere has a single tooth at its apex as in Fig. 15.

Plesiotype: Male, with associated pupal exuvium and cocoon, NW 23rd Ave. and NW 83rd St., Gainesville, Alachua Co., Florida, 2 February 1975 (Pinkovsky), to be deposited in the U.S. National Museum.

The wings of the female are about 4.5 mm long. The base of the radius is covered with setae and a basal cell is present on the wing. The frons bears short yellow hair. The pedisulcus is essentially absent and the prominent tooth on the basal claw has convex margins (Fig. 20). The apex of the abdomen is shiny. The genital fork has widespread narrow arms as in Fig. 21.

<u>Distribution</u>. Stone and Snoddy (1969) indicate *C. ornithophilia* occurs in Louisiana, Mississippi, South Carolina, Virginia, and Ontario. Tarshis and Stuht (1970) report this species from Maryland.

<u>Life History</u>. Stone and Snoddy (1969) mention that *C. ornithophilia*

Figure 20. Tarsal claw of a female of *C. ornithophilia*.

Figure 21. Genital fork and terminalia of a *C. ornithophilia* female.

is probably univoltine. Eggs collected in stream bottom samples and placed in laboratory rearing tanks yielded first instar larvae three days later (Tarshis, 1973). Larvae have been collected between 11 January and 18 March in South Carolina and adult females were reared on 6 February from pupae; females have been collected from 6 February to 1 May in Louisiana and Mississippi (Stone and Snoddy, 1969). Tarshis (1973) working in Maryland found larvae from 14 November to 29 April, pupae from 11 January to 22 April and adults from 2 to 17 March. Tarshis and Stuht (1970) captured adult *C. ornithophilia* between 28 February and 1 April in emergence cages over a stream in Maryland. When first to third instar larvae were collected and reared in the laboratory in aerated water at 15-22°C pupae appeared after 2 to 21 days and adults emerged 4 to 23 days after introduction of the larvae (Tarshis, 1973). Tarshis (1973) found adults emerged as soon as 2.5 hours after pupation [very difficult to believe] and that the number of males emerging exceeded the number of females emerging only on the first day.

Ecology. Tarshis and Stuht (1970) found massive numbers of larvae on leaves, rocks, and twigs in a stream that flowed .153 to 1.0 m/sec (.49-3.3 ft/sec) from a pond through an oak and willow woods. The stream was .92-1.53 m (3-5 ft) wide, 2.5-20 cm (1-8 in) deep and had a pH reaction between 6.9 and 7.4. In a similar stream that was slightly deeper and 1.22-3.66 m (4-12 ft) wide, larvae were collected at water temperatures of .5-15°C (Tarshis, 1973). Tarshis (1973) mentioned that pupae easily suffered injury and that development to adults did not occur when pupae were removed from their substrate. Larvae of *C. mutata*, *P. gibsoni*, *S. decorum*, *S. tuberosum*, *S. venustum*, and *S. vittatum* were collected with *C. ornithophilia* in a pond runoff stream where

C. ornithophilia predominated (Tarshis and Stuht, 1970).

Habits. Tarshis and Stuht (1970) suggest that *C. ornithophilia* oviposits on the stream surface and the eggs sink to the stream bed. Bennett (1960) in Ontario recovered *C. ornithophilia* (as *Cnephia* "U") from jays, hawks, robins, sparrows and other woodland birds. Tarshis (1972) fed *C. ornithophilia* on geese and ducklings in the laboratory and transmitted *L. simondi* to ducklings by the bite of the fly. Tarshis (1973) did not collect any *C. ornithophilia* from ducklings exposed on the banks of a stream containing larvae and pupae of the black fly and suggests that the target hosts were inappropriate or perhaps that the flies were absent at ground level and concentrated in the canopy.

Florida observations.

Stream Parameters

	Width	Depth	pH	Temperature		Velocity	
Mean:	2.57m	23.64 cm	4.7	14.7°C	(58.6°F)	.45 m/sec	(1.47 ft/sec)
Min:	.3	1.3	3.6	8.3	(47)	.23	(.75)
Max:	11	90	6.6	24.4	(76)	1	(3)

Cnephia ornithophilia is reported here for the first time from Florida. Both Stone (1965) and Stone and Snoddy (1969) mention that *C. pecuarum* occurs in Florida. Key characters, such as the shape of the tooth on the tarsal claw of the female, on specimens I obtained in Florida differed from those of *C. pecuarum*. In the U.S. National Museum I located one adult *Cnephia* specimen from Florida, listed for Site 204 below, which was a male that had been labeled as *C. pecuarum* but *C. ornithophilia* was penciled in underneath. Adult and immature specimens which I submitted from five collections representing two west Florida (Holmes and Liberty) and one east Florida (Alachua) counties were

identified by G.E. Shewell of the Canadian Biosystematics Research Institute as *C. ornithophilia*.

Cnephia ornithophilia was collected from 31 sites in 14 counties in Florida (Fig. 22). Most of the records represent collections of only a few, usually young larvae with small, pale histoblasts, but the larger overall body size compared to other species and distinctive head spots and gular notch are discernible even in young larvae. Larvae, pupae and associated adults were obtained at three sites — 28, 119 (Fig. 23), and 147). This species was first collected in the streams on 28 October and last collected on 18 April. Most collections occurred and the largest populations were encountered during December through March. Collections of *C. ornithophilia* at a number of sites were made during one winter month and the species was not found again until about the same time the following year. It appears that *C. ornithophilia* is univoltine in Florida. This species occurs in cool, fairly shallow flows, usually at most a few meters wide, most frequently with a velocity of .305-.61 m/sec (1-2 ft/sec) and a pH which normally is below 5. Immatures were also collected at Lebanon Station (Site 139) and Yellow Water Creek (Site 70) where the pH approaches neutrality. At most sites there was considerable vegetation especially trailing grass in the stream to which the immatures were attached. At Blues Creek (Site 2) and Site 28 sparce vegetation was present. Site 28 originated in a small swamp just upstream, was about .5 m wide, 10 cm or less deep and tumbled down a root-filled and rocky bed through woods. In January a heavy population of *C. ornithophilia* covered rocks, dead tree leaves, pine needles, and a few blades of trailing grass. Masses of 40 or more larvae encircled twigs about .6 cm in diameter in the current. Most of the

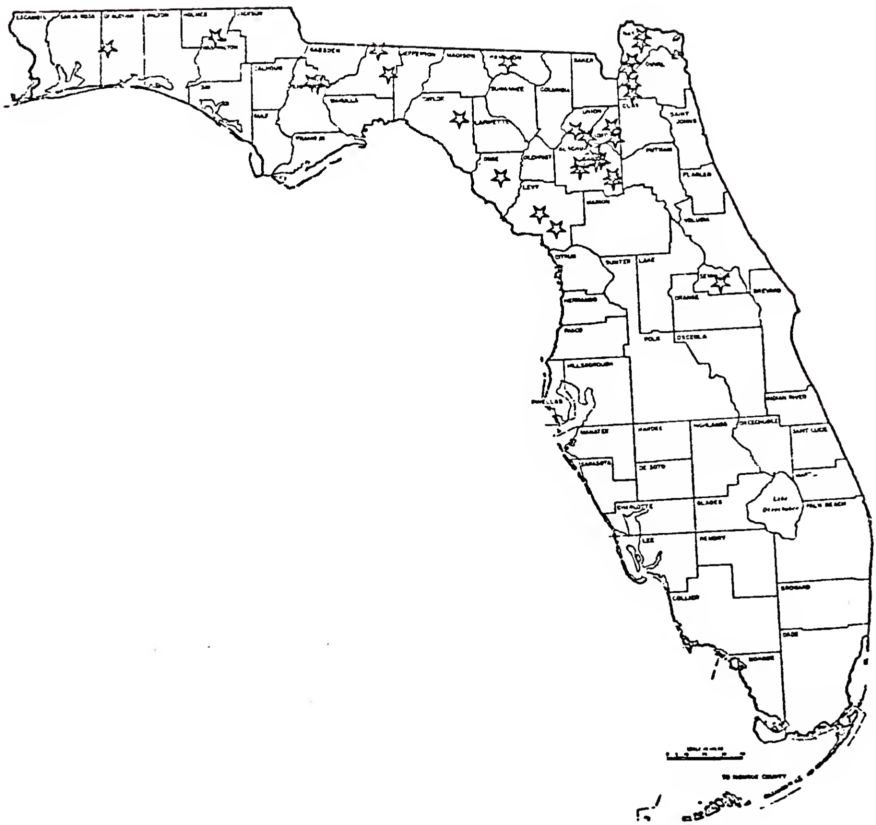

Figure 22. Collection locations for *C. ornithophilia* in Florida.

Figure 23. Site 119, Gum Creek, a stream inhabited by *C. ornithophilia.*

streams in which *C. ornithophilia* was located ceased flowing sometime during the year. At Site 99 at the Alapaha River *C. ornithophilia* was only collected when the river rose out of its normal channel and poured alongside the highway in flows into wooded areas.

In the streams with *C. ornithophilia* were found nine other black fly species (Table 3). Most frequently *S. slossonae* and *S. tuberosum*, both with 32 associations, were collected with *C. ornithophilia*. The next most common associates were *S. congareenarum* and *S. verecundum* which have seasonal occurrences that are similar to that of *C. ornithophilia*.

Swollen larvae infected with white masses of thousands of small white spheres were observed on at least five occasions. Usually these larvae were 4-6% of the total number present.

While dissecting adult females I observed that the salivary glands were large compared to those of *S. slossonae* and *S. congareenarum* and similar to the findings of Bennett (1963b), I found the glands lacked the golden pigment typical of most ornithophilic species. The eggs of *C. ornithophilia* were noted to be unusual also. Instead of being subtriangular as with the *Simulium* species the eggs of *C. ornithophilia* (.3 mm long and .1 mm wide) tapered to a point at both ends like a spindle. Immatures collected with their vegetation substrates from Site 119 on 31 December and reared in aerated water continued to yield emerging adults until 12 January. From larvae and pupae collected at Site 28 and reared in the laboratory 26 male and 25 female *C. ornithophilia* were obtained.

Adult females were captured in a Malaise trap at Site 2 at the IFAS Horticulture Unit during March. The record of Site 12 below is that of a *C. ornithophilia* female captured with a *S. slossonae* female feeding on

chickens. Six *C. ornithophilia* fed on turkeys in the lab but did not vector *L. smithi*. One female that emerged on 2 January was maintained on moist cotton and raisins in a holding carton and died 16 days later. Another female emerged on 4 January, fed on an infected turkey on 11 January, fed on clean birds on 14 and 19 January, and died on 25 January, 21 days after emergence. A third female fed twice on turkeys and died on 26 January or 22 days after she emerged.

Florida collection records for *C. ornithophilia*.

Alachua Co. 1) 1976: 21 Jan (S). 2) 1974: 2 Jan (S,M,L), 14 Dec (S); 1975: 10 Jan (S), 19-20 and 21-23 March (A — G.B. Fairchild). 6) 1974: 2 Jan (S,M), 7 March (S). 8) 1975: 10 Jan (S,M). 12) 1976: 22 Feb (A — H. Davis). 14) 1974: 7 March (S). 21) 1974: 2 March (S,M); 1976: 21 Jan (S,M). 23) 1975: 24 Jan (S). 28) 1975: 28 Oct (S), 9 Nov (S); 1976: 30 Jan (S,M,L,P,A).

Bradford Co. 43) 1975: 12 Jan (L); 1976: 31 Jan (S,M). 44) 1974: 4 Jan (S); 1976: 31 Jan (S). 48) 1975: 26 Jan (S,M).

Dixie Co. 68) 1975: 17 Jan (S).

Duval Co. 70) 1974: 4 Jan (M,L,P); 1976: 14 Feb (S,P). 72) 1974: 5 Jan (M); 1976: 14 Feb (S).

Hamilton Co. 99) 1975: 1 Feb (S,M); 1976: 14 Feb (S).

Holmes Co. 119) 1975: 19 Jan (S,M,L,P), 28 March (S,M,P,A), 30 Dec (S,M,L,P,A); 1976: 18 April (M).

Leon Co. 131) 1973: 17 Dec (S,M,L); 1975: 17 Jan (S), 29 Dec (S). 134) 1973: 19 Dec (S).

Levy Co. 135) 1973: 29 Dec (S). 139) 1975: 21 Dec (M,L).

Liberty Co. 142) 1975: 18 Jan (M). 147) 1973: 18 Dec (S); 1975:

18 Jan (S,M,L,P,A), 30 Dec (P).

Nassau Co. 159) 1974: 4 Jan (S,M,L); 1975: 12 March (M). 160) 1975: 12 March (M). 161) 1976: 14 Feb (L). 162) 1975: 12 March (P); 1976: 14 Feb (S,M).

Okaloosa Co. 168) 1973: 13 March (S,L — K. Tennessen).

Seminole Co. 204) 1960: 16 Nov (A — E.G. Jay).

Taylor Co. 211) 1975: 17 Jan (M).

Union Co. 217) 1974: 5 Jan (M).

Simulium (Byssodon) meridionale Riley

Simulium meridionale Riley,1887, Rep. of Entomol., U.S. Dep. Agr. Rep. for 1886: 513 (female).

Simulium occidentale Townsend, 1891, Psyche 6: 107 (female).

Simulium tamulipense Townsend, 1897, J. N.Y. Entomol. Soc. 5: 171 (female).

Simulium forbesi Malloch, 1914, U.S. Dep. Agr., Bur. Entomol. Tech. Ser. 26: 63 (female, male, pupa).

Simulium meridionale — Stone and Snoddy, 1969, Auburn Univ. Agr. Exp. Sta. Bull. 390: 28 (female, male, pupa, larva).

Taxonomy. Of the larva, pupa, male, and female in the original description by Riley (1887) only the female was not misidentified (Stone and Snoddy, 1969). Dyar and Shannon (1927) indicate the type locality for *S. meridionale* is probably Lake View, Mississippi and that type material is located in the U.S. National Museum. Stone and Snoddy (1969) mention that *S. meridionale* as recognized in the United States may actually refer to a group of sibling species.

Description. The larva is described by Stone and Snoddy (1969) as possessing large ventral tubercles, antennae which lack hyaline bands, and respiratory histoblasts which in the normal coiled condition are only slightly concave along the posterior border. The cephalic apotome bears a central posterior light spot surrounded by a fulvous ring with two dark pigmented rays projecting and diverging anteriorly.

The pupa is about 3 mm long and is situated in a well-constructed, slipper-shaped cocoon that projects forward noticeably along the ventral half. The respiratory organs each consist of about 25 filaments (Fig. 24).

The male as described by Malloch (1914) (as *S. forbesi*) and Stone and Snoddy (1969) is 2-2.5 mm long and possesses a velvety black to dark reddish brown scutum which lacks silvery spots. The abdomen is velvety black with a yellowish venter. The ventral plate is broad in ventral view, lacks marginal denticles and is not deeply notched. In end view the ventral plate displays only a very small median lobe.

The female wing is 2-2.5 mm long. The female is gray in appearance with a gray, pollinose frons and a gray scutum with three dark longitudinal lines (Fig. 25). The fore coxae are dark. The genital fork has a fairly thick stem and wide arms with dark, prominent, apical, dorsal projections (Fig. 17).

Distribution. Dyar and Shannon (1927) listed *S. meridionale* (as *S. occidentale*) from Jacksonville, Florida. Stone (1952) reported *S. meridionale* from a few sites in Alaska. Shewell (1957) recorded *S. meridionale* as an Austral and southern Boreal species from the northern transition regions of Canada. Snow et al. (1958) captured one female *S. meridionale* at Sugar Tree, Tennessee. Stone and Snoddy (1969)

Figure 24. The pupa and cocoon of *S. meridionale*.

Figure 25. The scutum of a *S. meridionale* female.

indicate the distribution is from Alaska to Indiana and south to California, Florida, and Mexico.

Life History. Stone and Snoddy (1969) indicate that the eggs of *S. meridionale* overwinter. Anderson and Dicke (1960) found adults could be collected from late May to late October and suggest there are at least four generations each year. Larvae matured in three weeks and pupae matured in three to four days in water temperatures 20°-24.4°C (68°-76°F) (Anderson and Dicke, 1960). Stone and Snoddy (1969) report at least four generations a year in Alabama with adults being collected from 16 March to 24 December.

Ecology. Shewell (1958) stated that *S. meridionale* is clearly a big river species. Tucker (1920) reported that adults appeared during times of high water and overflows of the Mississippi River. Townsend (1891) reported that *S. meridionale* (as *S. occidentale*) adults appeared as the Rio Grande River rose during May and June. Anderson and Dicke (1960) found immature stages in the Mississippi and Wisconsin rivers commonly attached to the sides of small rocks or beneath grass blades 5-15 cm (2-6 in) deep in currents usually less than .3 m/sec (1 ft/sec).

Habits. Anderson and Dicke (1960) collected adults up to 24 km (15 mi) from known breeding areas. The common name of *S. meridionale* is the turkey gnat (Blickenstaff, 1970). Anderson and DeFoliart (1961) found *S. meridionale* fed on white and bronze turkeys, chickens, pheasants, doves, and starlings with more flies being attracted to caged avian hosts placed at the tree canopy level than at ground level. Skidmore (1931) showed that *S. meridionale* was a vector of *L. smithi* to turkeys. Edgar (1953) reported a serious decline in egg production in chickens troubled by a spring outbreak of *S. meridionale* in Alabama.

Simulium meridionale was reported heavy and damaging to chickens in five Alabama counties in 1976 (U.S.D.A., 1976). Anderson et al. (1961) recovered eastern encephalitis virus from a pool of unengorged *S. meridionale* in a turkey brooder house and suggested that this species might serve as a biological vector. Townsend (1891), Anderson and DeFoliart (1961) and DeFoliart and Rao (1965) report that *S. meridionale* feeds on man. DeFoliart and Rao (1965) observed a greater tendency in *S. meridionale* to bite man after the flies had been exposed to a period of cool temperatures and suggested that *S. meridionale* might transmit encephalitis to humans.

Florida observations. Based primarily on adult collections, *S. meridionale* is recorded from 10 locations in 5 Florida counties (Fig. 26). The single record from Duval County refers to specimens collected years ago in Jacksonville by Mrs. A.T. Slosson and observed at the U.S. National Museum. Adults have been captured on the wing from 15 April to 26 May in counties bordering the Apalachicola River which is the only currently known breeding location of the species in Florida. Pupae of *S. meridionale* and two young, not positively identifiable larvae were collected on 17 April 1976 along the Apalachicola River at Richbourg's Landing (Site 145). The river was just beginning to recede from its spring flooding and had dropped about 1.83 m (6 ft) exposing rocks, some roots and sparse vegetation. At the collection site steep 9 m (30 ft) high banks of crumbling soil and rock were separated from the river by a strip of concrete and pebbles a few meters wide. A thick root sticking up vertically a short distance out of the water was retrieved and immatures were found on it from the apex to 55 cm (almost 2 ft) down. One large clump (1.8 x .8 m) of green grass-like vegetation with blades 15 cm (6 in) long and .9 cm (about 3/8 in) wide that was exposed on the

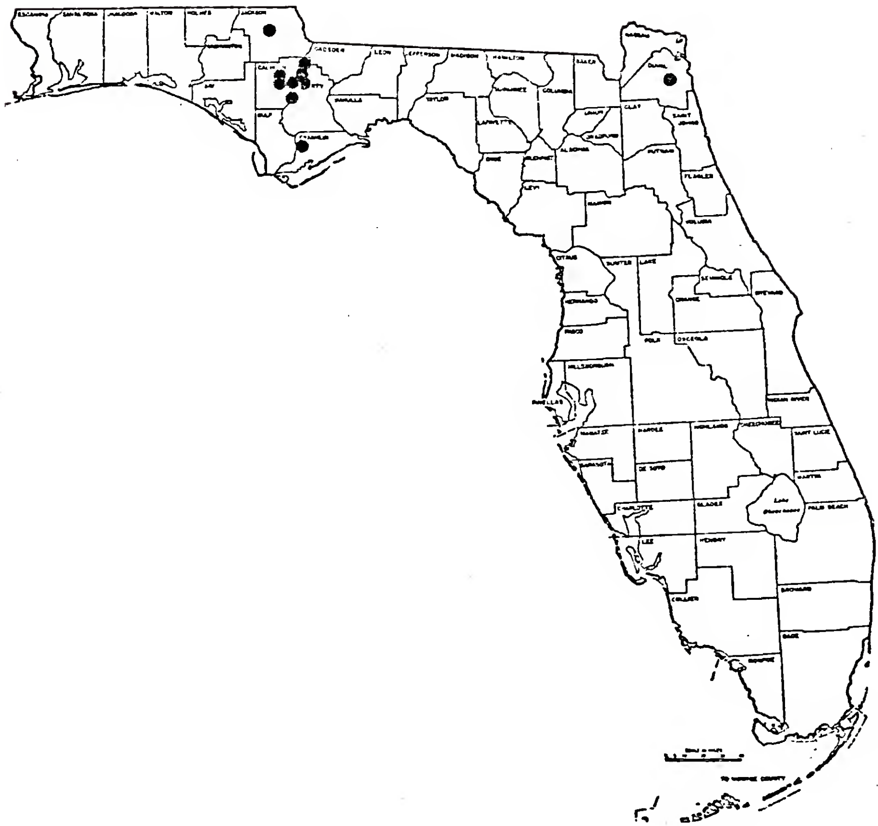

Figure 26. Collection locations for *S. meridionale* in Florida.

bank about 1 m out of the water harbored a large population of, mainly, *S. meridionale* pupae as did one small exposed bush. Other trailing tree roots yielded immatures but none were found on the pebbles or concrete in or out of the flow. The river was silty yellow brown, extremely wide and apparently very deep beyond the concrete strip. The current was about .61 m/sec (2 ft/sec), the water temperature was 21.5°C (70.5°F) and the pH was 6.5. Collected with the *S. meridionale* immatures were pupae of *S. jonesi*. Some adult *S. meridionale* were observed flying about the collection area. Only pupae and adult females of *S. meridionale* have been collected in this research.

The record for Site 53 is for *S. meridionale* adults at Blountstown which were reported to be causing deaths to young chicks. A number of reports of poultry and human suffering were received during collecting trips in Liberty and Calhoun counties during the April 1975 and 1976 *S. meridionale* mass emergences. Severe swelling and pain reactions to the bites of the bull flies, as the *S. meridionale* adults were called, were reported. The author found having twenty to thirty flies circling around the head, entering one's ears and crawling on one's neck and clothes slightly annoying, but he was never bitten.

The collection records indicate a single generation for this species in Florida. On dissecting wild-caught *S. meridionale* females, in the ovarioles next to well-developed eggs were noted small bodies which may have been small, developing eggs, possibly an indication of two ovarian cycles.

One *S. meridionale* which fed on a turkey infected with *L. smithi* was observed to feed on a clean turkey three days later. The clean bird became positive for *L. smithi* thus incriminating *S. meridionale* as a

vector of the disease in Florida. Wild caught flies were kept alive in the lab on water and raisins for a maximum of ten days.

Florida collection records for *S. meridionale*.

Calhoun Co. 51) 1975: 19 April (A). 53) 1973: 10 May (A). 52) 1975: 19 April (A).

Duval Co. 71) (no date): (A — Mrs. A.T. Slosson).

Franklin Co. 80) 1976: 2-3 April (A — G.B. Fairchild).

Jackson Co. 122) 1973: 26 May (A — W.W. Wirth).

Liberty Co. 145) 1976: 17 April (P,C). 149) 1975: 19 April (A). 150) 1975: 19 April (A); 1976: 17 April (A). 154) 1957: 15 April (A — F.S. Blanton); 1966: 20 May (A — H.V. Weems).

Simulium (Byssodon) slossonae Dyar and Shannon

Simulium slossonae Dyar and Shannon, 1927, U.S. Nat. Mus. Proc. 69(10): 34 (female, male).

Simulium slossonae — Underhill, 1944, Va. Agr. Exp. Sta. Bull. 94: 21 (female, male, pupa, larva).

Simulium slossonae — Stone and Snoddy, 1969, Auburn Univ. Agr. Exp. Sta. Bull. 390: 29 (female, male, pupa, larva).

Taxonomy. Dyar and Shannon (1927) briefly described the female of *S. slossonae* and also the male genitalia. The type locality is given as Biscayne Bay, Florida. The type male and seven paratype females were deposited in the U.S. National Museum (Dyar and Shannon, 1927). Underhill (1944) provided more detailed descriptions of the larva, pupa, male and female.

Description. The larvae are 4.5 to 5 mm long with reddish-brown

mottling and banding on the abdomen and thoracic region. The head spots are dark and distinct on a more pale head capsule. The posterior-lateral pair of spots on both sides are distinctly curved (Fig. 9). The gular notch is elongate, widest at three-fourths the way along its length anteriorly and tapers to a broad point (Fig. 27). There are 48 to 56 rays in each cephalic fan. The anterior arms of the anal cross-piece appear long. The anal lobes are large and cone-shaped.

The pupa is about 2.5 mm long in a slipper-shaped cocoon with distinctly concave anterior margins and a short, broad, anterior, mid-dorsal projection. The six, slender respiratory filaments arise in three pairs, lie close to each other, and have long petioles one-fifth to one-fourth the length of the filaments (Fig. 28).

The male is about 2.5 mm long and possesses a dark gray shiny scutum with black velvety areas centrally across the scutum which form an anterior triangle. The ventral plate of the male in end view is broadly rounded; the distimere is curved inward, rather flat and bears a lobe with hairs, basally (Fig. 29).

The female is about 2.5 mm long and is shining black with a shiny black frons. The abdomen bears three wide velvety black bands dorsally on the first few abdominal segments and more distally displays glistening black tergites. The female lacks setae on the underside of the subcosta and the base of the radius is bare dorsally. Each tarsal claw has a prominent basal tooth. The genital fork has thick arms with ventral projections which curve in toward each other forming a space like a U with a constricted opening (Fig. 30).

Distribution. Dyar and Shannon (1927) list *S. slossonae* from Biscayne Bay, Florida, and South Carolina. Underhill (1944) found this

Figure 27. Gular notch of a *S. slossonae* larva.

Figure 28. The pupa and cocoon of *S. slossonae*.

Figure 29. Terminalia of a male of *S. slossonae*.

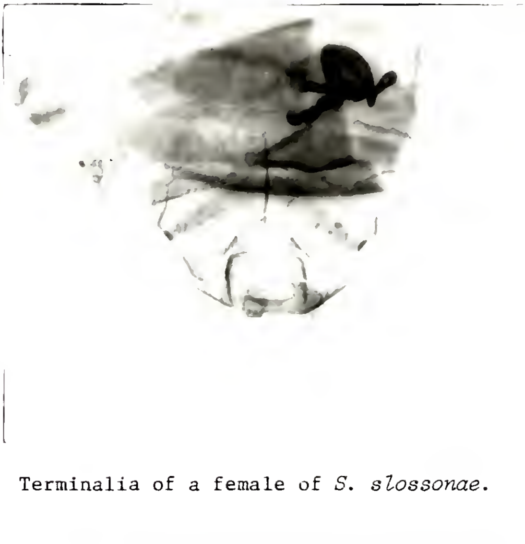

Figure 30. Terminalia of a female of *S. slossonae*.

species in the tidewater section of Virginia. Snow et al. (1958) do not list *S. slossonae* among the species occurring in the Tennessee River Valley. Stone and Snoddy (1969) list the additional states of Alabama, North Carolina, Georgia, Mississippi, and Texas for the distribution of *S. slossonae*.

Life Cycle. Larvae, pupae, and adults were collected as early as February in Jasper County, South Carolina (Jones and Richey, 1956). In Virginia *S. slossonae* was first found in collections made during March and last found during November (Underhill, 1944). Garris et al. (1975) found immatures of *S. slossonae* throughout a May through October collecting period. Jones and Richey (1956) concluded that the life cycle of *S. slossonae* was shorter than one month based on the discovery of many larvae and pupae, eight and twenty-one days, respectively, after a rain stimulated flow in streams previously devoid of black flies. Garris et al. (1975) state that *S. slossonae* is a multivoltine species present all year. In Alabama it was found from January through December except during June and September (Stone and Snoddy, 1969).

Ecology. Underhill (1944) mentioned that streams in Virginia inhabited by *S. slossonae* were .92-3 m (3 to 10 ft) wide, 20.3-38 cm (8 to 15 in) deep, usually had sandy-mud bottoms and were slow flowing with the swiftest portions reaching about .46-.61 m/sec (1.5-2 ft/sec). Jones and Richey (1956) found larvae in partially shaded sections of streams on tape grass and record maximum populations of 26 larvae per 6.5 cm^2 (1 in^2) of leaf surface. Significant plant growth and shade were typical of streams containing *S. slossonae* in Virginia (Underhill, 1944). Stone and Snoddy (1969) state that *S. slossonae* occurs in the swamp rivers of the South.

Habits. Underhill (1944) reports that *S. slossonae* feeds on turkeys. Wehr (1953) first showed *S. slossonae* could transmit *L. smithi*, experimentally, by intramuscular injection. Jones and Richey (1956) succeeded in transmitting *L. smithi* to a previously uninfected turkey by the bite of a *S. slossonae* female that had fed five days earlier on an infected bird. In South Carolina *S. slossonae* was captured from turkeys and humans but did not bite humans (Jones and Richey, 1956). Noblet et al. (1975) found that peaks in the adult populations of *S. slossonae* occurred during late July, late September, and late October, that the numbers of *S. slossonae* feeding on turkeys declined in November, and that few adult *S. slossonae* were present from mid-November until mid-February. Moore and Noblet (1974) report that *S. slossonae* commonly flies four miles after engorgement usually following stream courses toward swamps and mention adults may travel up to eight miles.

Florida observations.

Stream Parameters

	Width	Depth	pH	Temperature		Velocity	
Mean:	3.47 m	31.68 cm	4.5	21.3°C	(70.3°F)	.44 m/sec	(1.45 ft/sec)
Min:	.076	1	3.5	8.9	(48)	.12	(.4)
Max:	31	200	6.9	28.9	(84)	1.02	(3.33)

Stone and Snoddy (1969) state that *S. slossonae* is abundant in Florida and Davis et al. (1957) mention it was the most prevalent species encountered in Florida. I found that *S. slossonae* occurs in some streams all year long in Florida. The collection records listed below indicate that *S. slossonae* is the most widely distributed species in the State and has been collected at one time or another from 113 sites in a total of 46 counties (Fig. 31). It occurs in many parts

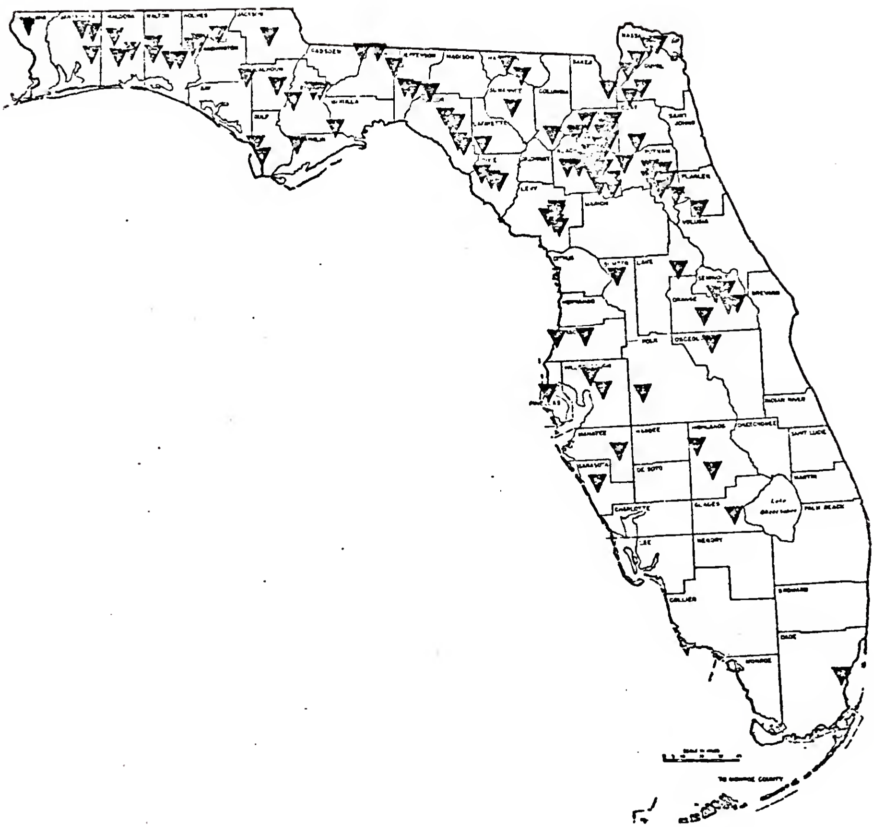

Figure 31. Collection locations for *S. slossonae* in Florida.

of west Florida, along the Gulf coast (Dixie, Levy Co.'s) where other species such as *S. tuberosum* are missing, is well established in north central Florida and is found as far south as just west of Lake Okeechobee. The most southern spot at which *S. slossonae* was found during this research was in Fisheating Creek which flows through cypress swamps in Glades County. The record from Dade County refers to material collected by Mrs. A.T. Slosson, probably around 1900 at a time when the Miami River had rapids. The distribution of *S. slossonae* most likely extends into other counties and a greater range will probably be demonstrated by later investigators.

As indicated by the stream parameters above *S. slossonae* was found in streams which on the average were fairly small and slow moving, with a mean velocity below .46 m/sec (1.5 ft/sec). These flows often exhibited a red brown or tea color and a low pH reaction in the vicinity of 4.4-4.5. In addition *S. slossonae* preferred streams with considerable stream vegetation and without excessive shading. Sites with most or all of these characteristics which were inhabited by *S. slossonae* most of the year and had large populations of immatures during certain periods included Sandy Hatchet Creek (Site 1), a tributary to Hatchet Creek (6), Site 8, Lochloosa Creek (21), Sites 43 (Fig. 32), 44, and 119, the stream from Lake Melrose (185) and Turkey Creek (216). Site 211 one mile west of the Fenholloway River is a location where *S. slossonae* was found year round in water of very low pH (3.5-4.0). Other streams (Sites 23 and 48) are very temporary in nature and clogged with grass and other green vegetation but when flowing in August and September contain good populations of *S. slossonae*. Immatures were found attached to all forms of aquatic vegetation including field and

Figure 32. Site 43, Double Run Creek, where *S. slossonae* immatures were collected.

eel grass, sedges, smartweed, pickerelweed, cattails, and *Hydrilla* as well as dead leaves, pine needles, twigs and, in lesser numbers, rocks and concrete chunks.

The size of *S. slossonae* populations seemed to be limited more by competition with other black fly species and actual flow stoppage than by increasing summer stream temperatures. Frequently, in streams that continued to run the largest numbers of immatures were located from April to November. In streams such as 43, 119, and 216, with an April to September period of maximum *S. slossonae* numbers, the initial increase in *S. slossonae* populations appeared to be a response to the increasing spring and early summer water temperatures and a lessening of substrate and perhaps food competition by winter and early spring species like *S. congareenarum* and *C. ornithophilia* as their populations began to decline.

While most streams from impounded water were inhabited by species such as *S. decorum* or *S. vittatum*, two sites were encountered where *S. slossonae* was the predominant species. At Site 177, a very intermittent flow below a lake in Pasco County, a tremendous population of *S. slossonae* including many pupae and cocoons was found during December on grass, twigs, dead leaves, and concrete. Site 85, a stream from Lake Melrose, flowed continuously and the smartweed, grasses and other vegetation and concrete chunks harbored good populations all year long.

S. slossonae occasionally appeared in streams with a neutral or slightly basic pH reaction (Sites 131, 213, 214) however population sizes did not approach those found in more typical *S. slossonae* streams.

S. slossonae was discovered rarely or only in small numbers in streams in Clay County (Sites 56 and 57) where *S. tuberosum* and *S. jonesi* predominated and a similar situation existed in Hatchet Creek (17) and

Sites 18 and 40 where pH and vegetation in most cases, were appropriate for *S. slossonae*. At Yellow Water Creek (70) *S. slossonae* was the predominant species in July and September of 1974 and from March through August 1975 maintained strong populations in coexistence with those of *S. tuberosum* and *S. jonesi*.

In the streams of Florida *S. slossonae* was associated at least once with eleven other black fly species (Table 3). *Simulium tuberosum* was most commonly found where *S. slossonae* occurred. Next frequently found was *S. jonesi* followed by *S. congareenarum*, *S. lakei*, and *S. verecundum*. *S. slossonae* was collected with *S. vittatum* on only fifteen occasions and never with *S. decorum* which reflects the more limited distribution of the latter species and generally different habitat preferences in *S. slossonae*.

No mermithids have been observed in the larvae of *S. slossonae* in Florida. Larvae have been observed swollen with white masses or infected with groups of tiny white spheres, probably microsporidian infections, on a number of occasions at just over a dozen sites. Infections were rarely found in more than 6 or 7 larvae per collection. In samples of 20 or more larvae of the stage infected infection rates ranged from 1.1% (1 of 90) to 7.7% (7 of 91). At Sites 21 and 186 1 of 14 and 1 of 4 medium-aged larvae were infected, respectively. At Sites 119 and 216 large, mature larvae were found with infected abdomens.

Adult females were captured in six counties during January through October in Manitoba traps (Table 4). Captures were made from dawn to dusk in winds from 0-16 km/hr, temperatures from 19.4-34°C and relative humidities from 49-100%. Females were also captured in Malaise traps (Site 25). Adults were observed to feed on domestic turkeys in the

field and in the laboratory during most of the year. *Simulium slossonae* has been shown to be the primary vector of *L. smithi* to turkeys in Florida (see *Leucocytozoon* transmission results section). The record for Site 12 below is that of *S. slossonae* captured while feeding on chickens. Adults have been netted from about the head and off the clothes of the author on a number of occasions even when temperatures in the field reached 32°C (90°F). Adults were netted during October at Sites 2 and 3 where *S. slossonae* immatures were never or infrequently found. One *S. slossonae* was captured biting a human on 5 July, 1972, in Gainesville, Florida (5). Katherine Sommerman collected an adult in Orlando in 1953 and labeled its actions as "probing".

<u>Florida collection records for *S. slossonae*.</u>

Alachua Co. <u>1</u>) 1973: 2 Nov (S,M,L,P), 14 Nov (S,M,L,P), 7 Dec (S,M,L,P), 13 Dec (S,M,L,P); 1974: 2 Feb (S,M,P), 7 March (S,M,P), 12 April (S,M,L,P), 5 May (M,P), 7 June (S,M,L,P,C), 6 July (S,M,L,P,C), 19 July (A), 20 July (S,M,L,P,C), 29 Aug (S,M,P,C), 28 Sept (S,M,P), 30 Oct (S,M,L,P), 23 Nov (S,M,L,P); 1975: 10 Jan (S,M,L), 12 Feb (M,P,A), 4 April (S,M,L,P), 10 May (P,C), 3 June (P,A), 21 June (S,M,L,P,C), 15 July (S,M,P,C), 25 Sept (S,M,C), 9 Nov (S,M,L,P, C,A); 1976: 21 Jan (S,M,L,P,A). <u>2</u>) 1974: 2 Jan (S), 17 Aug (M,P); 1975: 26 May (C), 3 Oct (A). <u>3</u>) 1975: 3 Oct (A). <u>5</u>) 1972: 5 July (A — D. Young). <u>6</u>) 1973: 22 Sept (S,P,A); 1974: 2 Jan (S,M), 2 Feb (S,L,P), 7 Mar (L), 2 April (S,M), 18 May (S,M,L,P,C), 30 June (S,M,L,P), 30 July (S,M,L,P), 14 Sept (S,L,P,C); 1975: 18 May (S,M), 16 Aug (S,M,L,P,C), 8 Oct (S,M,L,P,C), 6 Dec (S,M, L,P,C). <u>8</u>) 1973: 5 Sept (S), 2 Nov (L,P); 1974: 14 Feb (M), 2 April (S), 16 May (S,M,L,P), 30 June (S,M,L,P), 6 July (A),

3 Aug (S,M,L,P), 22 Sept (S,M,L,P,A), 25 Oct (S,M,L,P); 1975: 10 Jan (S,M), 12 Feb (S,M,L,P,C,A), 4 April (S,M,L), 18 May (P,C), 1 June (S,M,L,P,C,A), 18 June (S,M,C), 15 July (S,P), 25 July (P), 31 July (S,M,L,P,C), 30 Aug (S,M,P,C), 30 Sept (S,M,P,C,A), 9 Nov (S,M,A), 14 Dec (S,M,P); 1976: 6 March (S,M). 9) 1973: 5 Sept (S). 10) 1973: 5 Sept (S). 12) 1976: 22 Feb (A — H. Davis). 14) 1973: 5 Sept (S,M,L,P); 1974: 7 March (S), 30 May (S,M,L,P, C,A), 30 July (S,M,L,P), 14 Sept (S,M,L,P). 17) 1974: 1 July (P), 24 Aug (A); 1975: 1 March (S), 30 April (M). 18) 1974: 25 May (M), 24 Aug (M); 1975: 14 June (S), 17 July (A). 19) 1975: 17 July (S,M), 22 Oct (S). 20) 1974: 12 Jan (S), 6 July (S,M,P), 29 Aug (S,M); 1975: 18 April (M), 3 Oct (M,P,A). 21) 1974: 2 March (S,M,L,P), 11 April (S,M,L,P), 18 June (S), 28 June (S,M,L), 16 July (A), 30 July (S,M,L,P,C), 14 Sept (S,P); 1975: 24 Jan (S,L,P,A), 4 April (S,M), 27 Sept (S,M,L,P,C); 1976: 21 Jan (S,M, L,P,C), 29 Jan (M,P,A). 22) 1974: 28 June (A), 15 Aug (A), 28 Aug (S,M,L), 15 Sept (A), 7 Dec (S); 1975: 24 Jan (A), 29 April (S), 7 July (A), 5 Aug (S), 17 Oct (M,L,P); 1976: 6 March (S,A). 23) 1975: 24 Jan (S,M). 24) 1974: 25 May (M), 30 May (A), 28 June (A), 16 July (A), 24 July (A); 1975: 18 June (A). 25) 1975: 13 and 15 March (A — J. Glick). 28) 1975: 9 Nov (L); 1976: 26 March (A). 34) 1974: 19 July (A), 17 Aug (A). 36) 1974: 12 April (S,M), 5 May (C), 6 July (S,M,L,P), 24 Aug (S,M,P,C,A); 1975: 15 Jan (S,M,L,P,C), 4 April (S,M,L,P,A), 22 May (S,M), 22 Oct (S,M,L, P,C,A).

Bay Co. 40) 1975: 6 Sept (S).

Baker Co. 41) 1974: 4 Jan (S), 1 June (S,M,L,P,C), 17 Sept (S,M,L,P,C),

6 Nov (P); 1975: 1 Feb (S), 26 April (S,M,L,P,A), 21 June (S), 17 Aug (S,P), 14 Nov (S,L).

Bradford Co. 42) 1973: 6 Oct (S,M,L,P,C); 1974: 4 Jan (S), 20 July (S,M,P,C), 31 Aug (S,M,L,C), 9 Oct (S,M,L,P,C), 16 Nov (P,C); 1975: 26 Jan (S,C), 15 Sept (S,M,L,P,C), 19 Nov (S,M,L,P,C,A). 43) 1973: 17 Nov (S,P); 1974: 6 June (P,C), 20 July (S,M,L,P,C), 31 Aug (S,M,L,P,C), 19 Oct (S,M,P,C), 16 Nov (S,M,L,P); 1975: 12 Jan (S,M,L,P), 26 Jan (A), 5 April (S,M,P,C), 6 May (A), 1 July (S,M,L, P,A), 17 Aug (S,M,L,P,C,A), 26 Sept (S,M,L,P,C,A), 14 Nov (S,M,L,P,C); 1976: 7 Jan (S,M,L,P,C,A), 31 Jan (S,M,P,C,A), 24 March (A). 44) 1974: 4 Jan (S), 23 Feb (C), 14 April (S,M,L,P), 10 July (S), 20 July (S,M,L,P), 31 Aug (S,M,L,P,C,A), 9 Oct (S,M,L,P,C); 1975: 12 Jan (S,M,L,P,A), 5 April (S,M,L,P,C), 22 May (S,C), 17 July (S,M,L, P,C,A), 26 Sept (S,M,L,P,C,A), 19 Nov (S,M,L,P,C,A); 1976: 31 Jan (S). 46) 1975: 26 Sept (S,M,L,P,C). 47) 1974: 20 July (S,P,C), 21 Sept (S,M,L,P,C); 1975: 26 Jan (S), 26 April (S,M,L,P). 48) 1974: 3 Aug (S,M,L,P,A), 21 Sept (M,L,P,A); 1975: 5 April (P).

Calhoun Co. 50) 1975: 19 April (A).

Clay Co. 56) 1975: 2 Aug (S,P).

Columbia Co. 58) 1974: 21 Sept (S,P).

Dade Co. 63) (no date) (A — Mrs. Slosson).

Dixie Co. 67) 1974: 5 Aug (S). 68) 1975: 17 Jan (P,A), 26 March (S,M,P,C), 23 Aug (S,L,P,C). 69) 1975: 23 Aug (S).

Duval Co. 70) 1974: 4 Jan (S,M), 20 April (S,M,L,P,C,A), 10 July (S,M,P,A), 21 Sept (S,M,L,P,C); 1975: 12 March (S,M,L,P), 4 May (S,M,P,C), 1 July (S,M), 17 Aug (S,M,L,P,C,A); 1976: 14 Feb (S,M, L,P). 72) 1974: 5 Jan (M), 20 April (M,L,P,A), 10 July (S); 1975:

12 March (S,M,L,P,A), 4 May (S,M,L,P,C), 1 July (S,M,L,P,C,A); 1976: 14 Feb (P).

Escambia Co. 74) 1974: 16 June (S,M), 14 Oct (S).

Flagler Co. 77) 1974: 26 Jan (S), 25 May (S), 21 Aug (S,M,L,P,C,A), 7 Dec (P); 1975: 5 April (S,M), 9 July (S,M,P), 31 Oct (S,M,P,C).

Franklin Co. 80) 1976: 2-3 April (A — G.B. Fairchild).

Gadsden Co. 81) 1974: 15 June (L).

Glades Co. 95) 1974: 11-12 July (A), 26-27 July (S,M,L,C,A); 1975: 17 June (S,M,P,C), 27-28 June (A), 28 July (P,C), 29-30 July (A), 9-10 Aug (S,L,P,C,A), 17-18 Sept (A), 15-16 Oct (A).

Gulf Co. 97) 1970: 1-3 May (A — W.W.Wirth). 98) 1970: 3 May (A — W.W. Wirth).

Hamilton Co. 99) 1975: 1 Feb (A), 26 April (S,L). 101) 1975: 26 April (S,M,P,C,A), 3 Aug (S,P,C,A).

Highlands Co. 108) 1973: (S,P). 110) 1948: 13 July (A — R.H. Beamer); 1949: Jan (A — J.G. Needham); 1958: 26 Dec (A — S.W. Frost); 1959: 10 Jan (A — S.W.Frost), 30 March (A — S.W. Frost), 7 and 18 Nov (A — S.W. Frost); 1960: 23-24 Feb (A — S.W. Frost); 1961: 6 and 17 Feb (A — S.W. Frost).

Hillsborough Co. 112) 1964: I-III (A — J. Cross). 114) 1975: 12 Sept (P).

Holmes Co. 116) 1975: 28 March (A). 117) 1975: 11 June (M,P,C), 6 Sept (S,M,P,C), 30 Dec (S,C). 119) 1974: 15 June (S,M,L,P); 1975: 28 March (S), 11 June (S,M,L,P,C), 6 Sept (S,M,P,C), 30 Dec (M,P,C,A); 1976: 18 April (S,M,L,P,C).

Jackson Co. 123) 1939: 9 July (A — D.E.Hardy).

Jefferson Co. 124) 1975: 26 March (A). 125) 1975: 26 March (A), 23 Aug (S).

Lafayette Co. 130) 1974: 14 June (P); 1975: 26 March (A).

Lake Co. 170) 21 March (S), 3 Sept (P).

Leon Co. 131) 1974: 16 March (S,L); 1975: 26 March (L,P,A), 10 June (P,C), 23 Aug (P,C,A), 29 Dec (S). 134) 1973: 19 Dec (P).

Levy Co. 135) 1973: 7 Oct (P); 1974: 2 March (S,M,L), 3 Aug (S,M,L,P, C,A), 15 Sept (S,M); 1975: 31 Jan (S,M), 23 March (P,A), 17 May (P), 30 Oct (S). 136) 1974: 15 Sept (S). 137) 1974: 2 March (S), 18 June (S), 3 Aug (S,M), 15 Sept (S,M,L,P); 1975: 23 March (S). 138) 1975: 31 Jan (S,M,P,C), 30 Oct (S,A). 139) 1975: 23 March (S,P,C), 11 Sept (S,M,L), 21 Dec (S). 141) 1975: 8 July (P), 30 Oct (S,M).

Liberty Co. 142) 1975: 18 Jan (S,M), 24 Aug (S). 143) 1976: 1-2 April (A — G.B. Fairchild). 147) 1975: 24 Aug (M,P).

Madison Co. 153) 1974: 16 March (S,M,L,P), 5 Aug (S,M,L), 12 Oct (S,M,L,P,A); 1975: 17 Jan (S,M,L), 26 March (S,M,L,P,C,A), 23 Aug (S,M,L,P,C,A), 29 Dec (S,M).

Manatee Co. 156) 1975: 12 Sept (S).

Nassau Co. 159) 1974: 4 Jan (S,P,C), 20 April (S,M,L,P,A), 10 July (S,M,L), 24 Aug (S,M,L,P,C,A), 9 Oct (S,P,C), 4 Dec (C); 1975: 12 March (S,M,L,P,A), 4 May (S,M,P,C), 1 July (S,M,L,P), 26 Sept (S,M,L,P,C); 1976: 14 Feb (S,M,P,A). 160) 1974: 20 April (S,P,A), 10 July (S), 24 Aug (S,M), 9 Oct (S,M,L,P,C); 1975: 12 March (S), 4 May (S,M,P,A), 1 July (C), 26 Sept (S,M,C). 161) 1974: 20 April (M,P,C,A), 10 July (S,M), 24 Aug (S,M,L,P), 9 Oct (S,A); 1975: 12 March (S,M), 4 May (A), 1 July (S), 26 Sept (S,P); 1976: 14 Feb (C). 162) 1974: 20 April (M,L,P,C), 10 July (L,P,C), 24 Aug (S,M,P), 9 Oct (S,M); 1975: 12 March (S,M,L,P,C), 4 May (A), 22 July (S,M, L,P,C).

Okaloosa Co. <u>165</u>) 1974: 18 March (M,L,P,A), 16 June (S,M,P), 7 Aug (S,M,L,P), 14 Oct (S,M,L,P); 1975: 28 March (S,L), 12 June (S,M). <u>166</u>) 1974: 18 March (C), 14 Oct (S); 1975: 6 Sept (S). <u>168</u>) 1975: 12 June (S,P), 6 Sept (S,C).

Orange Co. <u>169</u>) 1973: 11 Sept (S,P); 1974: 13 July (S,M,L,P); 1975: 15 March (P), 4 July (S,P), 30 Oct (S,M,L,P,C). <u>174</u>) 1936: 31 March (L,P — U.S.N.M.); 1941: 13 March (L,P,A — W.V.King); 1947: 28 Jan (P,A — H.K. Gouck), 5 and 11 Feb (P,A — H.K. Gouck), 10 March (A — H.K. Gouck), 13 March (P — H.K. Gouck); 1953: 5 Aug (A — K.M. Sommerman), 4 Oct (A — K.M. Sommerman). <u>176</u>) 1936: 31 March (A).

Pasco Co. <u>177</u>) 1975: 23 March (P,C,A), 21 Dec (S,M,L,P,C,A). <u>178</u>) 1939: 13 July (A — D.E. Hardy).

Osceola Co. <u>179</u>) 1932: 1 Feb (A — A.L. Melander).

Pinelas Co. <u>180</u>) 1936: 5 March (A — collector unknown).

Polk Co. <u>182</u>) 1975: 27 May (S,M).

Putnam Co. <u>185</u>) 1973: 17 Nov (S,M,L,P,C,A); 1974: 5 Jan (S,M,L,P,A), 14 Feb (S,M,L,P), 26 March (S,M,L,P,C), 18 May (S,M,L,P,C,A), 6 July (S,M,L,P,A), 12 Aug (S,M,L,P,C,A), 6 Oct (S,M,L,P,A), 23 Nov (S,M, L,P,C); 1975: 15 Jan (S,M,L,P,C,A), 4 April (S,M,L,P,C), 11 May (S,M,L,P,C), 14 June (S,M,L,P), 17 July (S,M,L,P,C,A), 30 Sept (S,M,L,P,C,A), 14 Dec (S,M,L,P,C,A). <u>186</u>) 1974: 19 Jan (S,M,L,P,A), 14 April (S,M,L,P,C), 25 May (S,M,L,P), 6 July (S,M,L,P,C,A), 17 Aug (S,M,L,P,C), 6 Oct (S,M,L,P,C,A), 23 Nov (S,M,L,P,C); 1975: 12 Feb (S,M,P,A), 18 April (S,M,P), 26 June (P,C), 25 Sept (S,M,L, P,C). <u>187</u>) 1974: 6 July (P). <u>188</u>) 1964: 9 April (A — H.A. Denmark). <u>190</u>) 1974: 7 Dec (S,M,C). <u>192</u>) 1974: 6 Oct (S), 7 Aug (C). <u>193</u>) 1974: 26 Jan (S,M,L,P,C,A).

Santa Rosa Co. 195) 1975: 12 June (P). 196) 1974: 16 June (S); 1975: 12 June (S). 197) 1973: 23 May (A — W.W. Wirth).

Sarasota Co. 198) 1967: 13 March (S,L — W. Beck).

Seminole Co. 203) 1974: 21 March (S,M,L,P), 12 May (S,M,P), 11 July (M,L,P), 3 Sept (S,M,L,P,C), 28 Nov (S,M); 1975: 15 March (L,P), 4 July (S,M,L,P,C), 31 Oct (S,M,L,P,C). 204) 1960: 15 and 31 March (Larvae — E.G. Jay).

Sumter Co. 206) 1973: 21 Nov (M,L,C); 1974: 28 Nov (S,M,P).

Suwanee Co. 209) 1945: 10 Jan (A — D.J. Taylor).

Taylor Co. 210) 1954: 8 April (P — C.M. Jones); 1974: 16 March (S,M, L), 14 June (S,M,L,P), 5 Aug (S,M,L,P), 12 Oct (S,M,L,P); 1975: 17 Jan (S,M,L,P), 26 March (S,M,L,P), 10 June (S,M,L,P,C), 23 Aug (S,M,L,P,C). 211) 1974: 16 March (S,L,P), 14 June (S,M,L,P), 5 Aug (S,M,P), 12 Oct (S,M,L,P); 1975: 17 Jan (S,M,L,P,C), 26 March (S,M,L,P,C,A), 10 June (S,M,L,P,C), 23 Aug (S,M,L,P,C), 29 Dec (P,C). 213) 1974: 14 June (S,M,L,P), 5 Aug (M,P), 12 Oct (P); 1975: 17 Jan (S,L), 23 Aug (S,P). 214) 1975: 17 Jan (S,M,L). 215) 1975: 26 March (S,L,P,C,A), 23 Aug (S,M,L,P,C).

Union Co. 216) 1974: 5 Jan (S), 23 Feb (S,M,L,P,C), 14 April (S,M,L,P,C), 1 June (S,M,L,P), 6 July (S,M,L,P), 24 Aug (S,M,L,P,C), 9 Oct (S,M,L,P,C), 6 Nov (S,M,L,P,C,A); 1975: 12 Jan (S,M), 5 April (S,M,L,P,C,A), 4 May (S,M,L,P,C,A), 6 May (A), 11 May (A), 1 June (S,M,L,P,C), 15 June (S,M,L,P,A), 22 July (S,M,L,P,C), 5 Oct (S,M, L,P,C), 14 Nov (S,M,L,P,C); 1976: 7 Jan (S,M,L,P,C,A), 17 Feb (S,M,L,P,C,A), 24 March (P,A). 217) 1974: 23 Feb (A), 6 July (S,P,C), 24 Aug (S,L,P,C), 9 Oct (S,M,P,C); 1975: 1 June (S,L,P,C), 22 July (P), 5 Oct (S,M).

Wakulla Co. 219) 1970: 29 April (A — W.W. Wirth).

Walton Co. 220) 1974: 13 Oct (L); 1975: 11 June (P,A). 221) 1975: 11 June (M), 6 Sept (M). 222) 1974: 6 Aug (S,M,A). 223) 1975: 28 March (S), 11 June (S,L).

Simulium (Eusimulium) congareenarum (Dyar and Shannon)

Eusimulium congareenarum Dyar and Shannon, 1927, Proc. U.S. Nat. Mus. 69(10): 20 (female).

Simulium congareenarum — Jamnback and Stone, 1957, Ann. Entomol. Soc. Amer. 50: 395 (male, female, larva, pupa).

Simulium congareenarum — Stone and Snoddy, 1969, Auburn Univ. Agr. Exp. Sta. Bull. 390: 27 (male, female, larva, pupa).

Taxonomy. Dyar and Shannon (1927) first described a female of *S. congareenarum* (as *Eusimulium congareenarum*) and the holotype location was given as Congaree, South Carolina. The holotype and twenty-three paratype females were deposited in the U.S. National Museum. Jamnback and Stone (1957) provided the first descriptions for the larva, pupa, and male of *S. congareenarum* and redescribed the female. Davies et al. (1962) mention that morphological and cytological evidence suggests that *S. congareenarum* is a species complex. Stone and Snoddy (1969) indicate that biological information on hand also suggests a complex exists.

Description. Mature larvae are 5.5-6 mm long with a yellow head capsule and dark brown head spots. The anterior medial group of spots is made up of 4 or 5 small distinct spots which are widely separated from the posterior medial group of spots (Fig. 33). Each cephalic fan

Figure 33. Cephalic apotome of a *S. congareenarum* larva.

Figure 34. Venter of the larval head capsule of *S. congareenarum*.

contains 57-61 rays. The gular notch is in the form of a small square or is of a shallow U-shape and extends less than one-fifth the distance to the submental teeth (Fig. 34). The abdomen is reddish brown with lighter intersegmental areas. The ventral, anal tubercles are large and cone-shaped.

The pupa is about 3 mm long and bears a pair of 12-filamented respiratory organs (Fig. 35). The anterior edges of the cocoon in a lateral view are concave and a long anterior projection occurs dorsally on the cocoon.

The wing of the male is 2.5 mm long. The scutum of the male is velvety black but is covered with many golden hairs. The scutellum bears long erect silvery or golden hairs. The base of the radius bears hairs dorsally. The ventral plate in end view is a good sized sharp-pointed V. The distimeres are only slightly curved and taper to a sharp point (Fig. 36).

The wing of the female is about 2.5 mm long. The frons and clypeus are gray pollinose in appearance. The scutum appears gray with many closely appressed pale hairs. The wings bear setae on the base of the radius and there is a small basal cell. The pedisulcus on the second tarsal segment is shallow. The tarsal claw has a prominent basal tooth with convex margins. The genital fork has stout arms and prominent inner projections (Fig. 37).

Distribution. In addition to its occurrence in South Carolina *S. congareenarum* has been reported from New York, Florida (Alachua Co.), Georgia, Louisiana, Maryland, and Virginia (Jamnback and Stone, 1957), Ontario, Canada (Davies et al., 1962), Connecticut (Stone, 1964), Alabama (Stone and Snoddy, 1969) and New Jersey (Crans and McCuiston, 1970a).

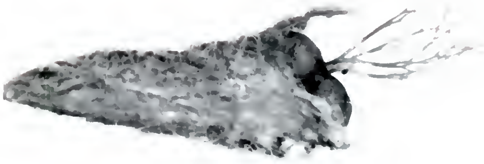

Figure 35. Pupa and cocoon of *S. congareenarum.*

Figure 36. Terminalia of a male of *S. congareenarum.*

Figure 37. Terminalia of a female of *S. congareenarum*.

Life history. In Canada overwintering eggs of the first generation begin hatching in early April, pupae and adults are present by May and a possible second generation is indicated by a peak of pupation during July (Davies et al., 1962). Stone (1964) reports that adults from the overwintering larvae emerge during early March in the southern regions and in April in the north. In South Carolina larvae of *S. congareenarum* were collected during January, were abundant in February and March and adults were present during March through early May (Jones and Richey 1956; Anthony and Jones, 1958; Noblet et al., 1972). Jones and Richey (1956) also report collecting a few larvae and pupae through 11 June and one larva on 13 July. In South Carolina *S. congareenarum* appears to be a univoltine species (Garris et al., 1975). A 20- to 25-day life cycle from egg to adult emergence is reported for *S. congareenarum* and other *Simulium* species near Pageland, South Carolina (Kissam et al., 1975).

Ecology. Jamnback and Stone (1957) collected *S. congareenarum* in a permanent creek .61 m (2 ft) wide and 15 cm (6 in)deep. Stone (1964) indicates that the immatures of *S. congareenarum* attach to vegetation in fairly slow-flowing permanent streams. Garris et al. (1975) indicate that this species usually occurs in swampy streams.

Habits. During early April *S. congareenarum* adults were collected from breeder turkeys (Anthony and Richey, 1958). The number of adults attracted to turkeys decreased to zero by mid-May (Jones and Richey, 1956). Noblet et al. (1972) found that the adults feed on turkeys, ducks, and chickens and showed, by injection of homogenized infected flies, that *S. congareenarum* was a vector of *L. smithi* to turkeys. Noblet et al. (1975) report that *S. congareenarum* is an important vector of *L. smithi* in the early spring.

Florida observations.

Stream Parameters

	Width	Depth	pH	Temperature	Velocity	
Mean:	2.55m	30.97 cm	4.3	16°C (61°F)	.46 m/sec	(1.51 ft/sec)
Min:	.076	1.27	3.5	6.7 (44)	.15	(.5)
Max:	11	166	6.7	26.1 (79)	1	(3.3)

Figure 38 shows the distribution of *S. congareenarum* in Florida where it was found in 16 counties at 34 sites. This species was only collected in the northern portion of the state. One young larva was collected on 6 September at Little Reedy Creek (Site 117) in west Florida and a large larva was recovered from Water Oak Creek (44) in east Florida on 26 September. At all other locations favorable for *S. congareenarum* larvae were not found in the fall before early October. Larvae and pupae were present in many streams in greatest numbers from January through March and occasionally into April. By May the numbers of *S. congareenarum* were significantly lower, both in permanent streams where *S. slossonae* also occurred and was building up its populations, and in temporary streams where the flow reached its lowest level or ceased prior to the summer rainy season. The latest record for larvae was 11 June at Site 117. Adults were captured during January, March, April and late May. It appears that *S. congareenarum* is able to complete at least two and possibly three generations each breeding season and usually spends the summer in the egg stage or possibly, in more permanent flows, as small numbers of larvae.

The streams in which the largest populations of *S. congareenarum* were encountered (8, 43, 44, 117, 131, 153, 211, 216 — Fig. 39) were usually 1 to 3 m wide 10 to 50 cm deep with currents .46 to .76 m/sec

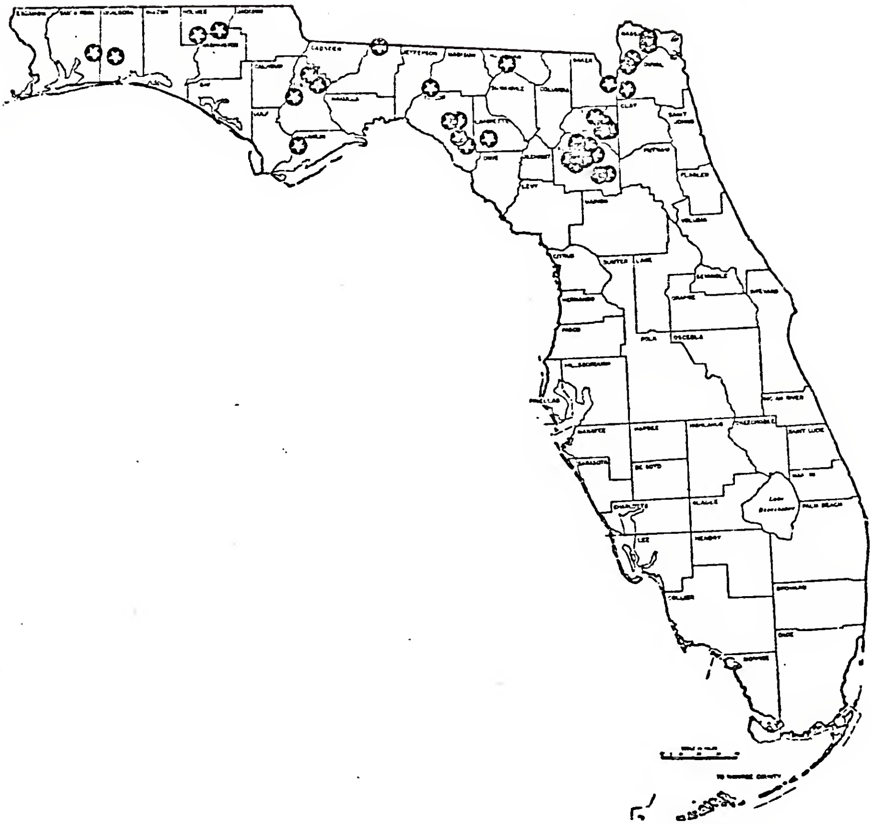

Figure 38. Collection locations for *S. congareenarum* in Florida.

Figure 39. Site 216, Turkey Creek, a typical *S. congareenarum* stream.

(1.5-2.5 ft/sec) and a low pH, 3.5-4.5. These streams had trailing grass as a primary type of vegetation in the flow and, generally, sandy bottoms. Larvae and pupae were found attached to grass, sedges, large and small-leafed aquatic vegetation, dead leaves, pine needles and, occasionally, rocks.

Simulium congareenarum was collected with nine other black fly species (Table 3). Most notably it occurred with *S. slossonae* and *S. tuberosum* and was recovered frequently with two other winter and early spring species, *C. ornithophilia* and *S. verecundum*.

Infected larvae with tiny white spheres in the abdomen and thorax or a large white mass in the abdomen were noted only a few times in collections of *S. congareenarum* and only involved 1-3 larvae per collection.

Adults reared in the laboratory during April 1975 and March - April 1976 which were dissected before taking a blood meal were found to have well-developed eggs in the ovaries. This suggests some females of *S. congareenarum* may be autogenous at least for the first ovarian cycle. It may also help explain the reluctance of reared *S. congareenarum* to feed on turkeys in the laboratory while wild-caught *S. congareenarum*, which may have already oviposited, fed more readily. Adults fed a number of times on turkeys in the field and more often in the laboratory and transmitted *L. smithi* to clean birds on three occasions. This is the second most important vector of *L. smithi* to turkeys in Florida. The vector potential of *S. congareenarum* is limited by its seasonality and the relatively lower number of adults in flight.

Adults were captured in Manitoba traps during January-March at four sites in four counties (Table 4), in a Malaise trap at Site 2 in March,

and from exposed turkeys on a few occasions during early spring (Table 8). *Simulium congareenarum* was easy to differentiate in the field from the frequently collected *S. slossonae* by the more robust appearance of the former, its more awkward ambling up a Manitoba canopy, and its silvery gray frons.

Florida collection records for *S. congareenarum*.

Alachua Co. 1) 1973: 7 Dec (L), 13 Dec (L); 1975: 12 Feb (S); 1976: 21 Jan (S). 2) 1975: 19-20 March (A — G.B. Fairchild). 6) 1974: 2 April (S); 1975: 8 Oct (M). 8) 1974: 14 Feb (S), 2 April (S), 16 May (S); 1975: 10 Jan (S,M), 12 Feb (M,P,C,A), 9 Nov (S), 14 Dec (S); 1976: 6 March (M). 14) 1974: 7 March (S), 30 May (S,L). 21) 1974: 2 March (S), 11 April (S,L); 1975: 4 April (M,C); 1976: 21 Jan (S,M), 29 Jan (S,P,A). 22) 1975: 24 Jan (A); 1976: 6 March (A). 23) 1975: 24 Jan (S,M). 36) 1975: 15 Jan (L), 4 April (M), 22 Oct (S). 38) 1957: (no month or stage — Jamnback and Stone).

Baker Co. 41) 1974: 4 Jan (S); 1975: 1 Feb (S).

Bradford Co. 42) 1974: 4 Jan (S). 43) 1973: 17 Nov (S,M,P,A); 1974: 4 Jan (S,M), 16 Nov (P,A); 1975: 12 Jan (S,M,L,P,C), 26 Jan (A), 5 April (S,M,L,C); 1976: 7 Jan (S,M,P,C,A), 31 Jan (S,M,L). 44) 1974: 4 Jan (S), 23 Feb (C), 14 April (S,M,L); 1975: 12 Jan (S,L), 5 April (S), 26 Sept (L); 1976: 31 Jan (S,M).

Duval Co. 70) 1974: 4 Jan (S,M,P); 1975: 12 March (S).

Franklin Co. 80) 1976: 2-3 April (A — G.B. Fairchild).

Hamilton Co. 99) 1976: 14 Feb (M).

Holmes Co. 117) 1975: 19 Jan (S,M,L,P), 28 March (S,M,L,P), 11 June (S,M), 6 Sept (M), 30 Dec (S,M). 119) 1975: 19 Jan (S,M,L,P), 28 March (S,M,L,P,A), 30 Dec (S,M,L,P,C); 1976: 18 April (S).

Lafayette Co. <u>130</u>) 1975: 17 Jan (S), 26 March (C).

Leon Co. <u>131</u>) 1973: 17 Dec (S,M,L); 1974: 16 March (S,M,L,P,C); 1975: 26 March (S,L), 29 Dec (S,M,L).

Liberty Co. <u>143</u>) 1976: 1-2 April (A — G.B. Fairchild). <u>144</u>) 1973: 18 Dec (S). <u>147</u> 1974: 17 March (S); 1975: 18 Jan (S,M), 27 March (S,M), 30 Dec (P,C,A).

Madison Co. <u>153</u>) 1974: 16 March (S,M,L,P,C), 12 Oct (M); 1975: 17 Jan (S,M,L,P,C), 26 March (S,M,L,P,C,A), 29 Dec (S,M,L).

Nassau Co. <u>159</u>) 1974: 4 Jan (S); 1975: 12 March (A); 1976: 14 Feb (S,M,L). <u>160</u>) 1974: 4 Jan (S), 20 April (S,M,L); 1975: 12 March (S,M). <u>161</u>) 1975: 12 March (S,M); 1976: 14 Feb (L). <u>162</u>) 1975: 12 March (P,A).

Okaloosa Co. <u>168</u>) 1973: 13 March (S,M,L,P — K. Tennessen).

Santa Rosa Co. <u>197</u>) 1973: 23 May (A — W.W. Wirth).

Taylor Co. <u>210</u>) 1974: 16 March (M); 1975: 17 Jan (S,M,L), 26 March (S,M,L,P), 28 Dec (S,M). <u>211</u>) 1974: 16 March (S,M,L,P); 1975: 17 Jan (S,M,L), 26 March (S,M,L,P), 29 Dec (P). <u>214</u>) 1975: 17 Jan (S,M,L,P). <u>215</u>) 1975: 26 March (S,M,L).

Union Co. <u>216</u>) 1974: 5 Jan (S,M,P), 23 Feb (S,M,L,P,C), 14 April (S,M,L,P), 9 Oct (S), 6 Nov (S,M); 1975: 12 Jan (S,M,L,P,A), 5 April (S,M,L,P,A), 4 May (S,M,C), 14 Nov (S); 1976: 7 Jan (S,M, L,P,A), 17 Feb (S,M,L,P,C,A), 24 March (P,A).

<u>*Simulium (Phosterodoros) dixiense* Stone and Snoddy</u>

Simulium dixiense Stone and Snoddy, 1969, Auburn Univ. Agr. Exp. Sta. Bull. 390: 34 (female, male, pupa).

Taxonomy. A female of *S. dixiense*, with the associated pupal exuvium and cocoon, was designated as the holotype (Stone and Snoddy, 1969). The type locality is Lewis Creek, Washington Co., Alabama. The holotype is in the U.S. National Museum.

Description. Stone and Snoddy (1969) indicated that unless the respiratory filaments of well-developed larvae were examined, they were probably not distinguishable from *S. jonesi* or *S. nyssa*. During this research *S. dixiense* was found to be readily distinguished from other species and had the following characteristics: the larva is 4.5 mm long; the color of the larval head capsule and abdomen is a striking light yellow with the larvae appearing almost clear and very difficult to detect on eel grass except for the dark histoblasts of well-developed larvae and the sclerotized anal X-piece and the mandibles; the head capsule is conspicuously elongate; the head spots are fairly faint and light brown in color and near pupation these spots disappear and are replaced by whitish yellow or colorless areas (Fig. 40); the gular notch is elongate, extends two-thirds the distance to the submental teeth, is widest about halfway along its length but not nearly as wide as long, and is pointed anteriorly (Fig. 41); the hypostomium bears a dark sclerotized band at the base of the teeth; the cephalic fans consist of 55-58 rays each; the shape of the fully open fan is more like an elongate kidney bean than a semi-circle; the antennae extend slightly beyond the cephalic fan stalk; the abdomen is swollen and curved posteriorly giving the anal hooks a more ventral than terminal placement; there are approximately 63 rows of anal hooks with 12 hooks in each row; the anal tubercles are prominent; the anterior arms of the anal X-piece appear long; and the body in alcohol is bowed or curved into a C-shape (Fig. 11).

Figure 40. Dorsal view of the head capsule of a *S. dixiense* larva.

Figure 41. Gular notch and hypostomium of a *S. dixiense* larva.

The pupa is 2.5-3 mm long with filaments 1 mm long. The respiratory organ consists of 10 filaments in 5 pairs. The fourth petiole from the dorsal is the thickest and longest extending 1/3 or more of the length of the filaments. The anterior margins of the cocoon in lateral view appear straight, not convex, and slope back from the ventral to the dorsal. There is a large aperture on each side of the cocoon anteriorly (Fig. 12). Pupae often appear bright yellow when freshly collected.

The male bears wings 2-2.5 mm long. The scutum has a pair of large silvery spots anterior laterally and a wide, posterior silvery area with the remainder of the scutum being velvety black and expanding anteriorly between the two spots. The abdomen is velvety brownish black with iridescent spots along the side. According to Stone and Snoddy (1969) the terminalia are not distinguishable from those of *S. jenningsi* (Fig. 42).

The female's wings are 2-2.5 mm long. The stem vein hairs are dark and there are no setae on the base of the radius. The frons is shiny dark brown. The scape and pedicel of the antennae are orangish-red and the flagellum is dark brown. The scutum is shiny dark brown with many small golden hairs and appears pollinose laterally. The legs are dark distally, lighter yellow or yellow brown proximally. The abdomen is velvety black except the last four tergites which are shiny dark brown. The stem of the genital fork is thin and dark while the arms are wider, lightly sclerotized and end in a dark sclerotized tip (Fig. 43).

Distribution. Stone and Snoddy (1969) report *S. dixiense* from Alabama and South Carolina.

Life history. Stone and Snoddy (1969) suggest this species is multivoltine and report collecting pupal exuviae in early March and

Figure 42. Male terminalia of *S. dixiense*.

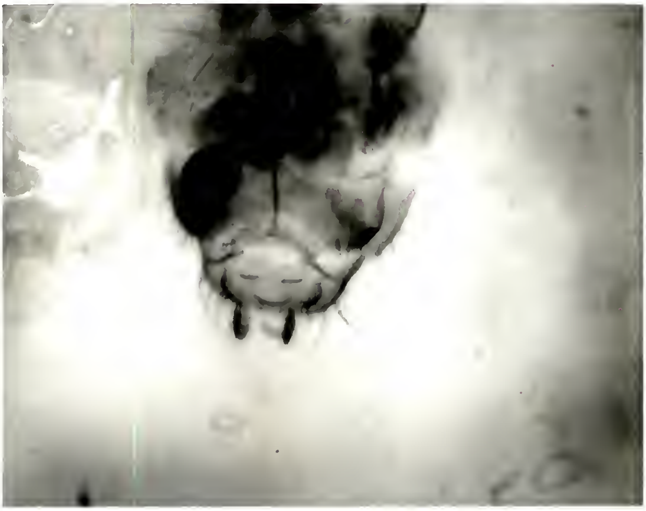

Figure 43. Terminalia of a female of *S. dixiense*.

larvae and pupae into November. Garris et al. (1975) in a survey conducted from May through October in Sumter County, South Carolina, collected pupae of *S. dixiense* only in May and July.

Ecology. Stone and Snoddy (1969) found that *S. dixiense* prefers the small swamp streams of the Southeast and collected this species in Alabama only from the Lower Coastal Plain. Larvae and pupae were usually collected in sand-bottomed streams 15-20 cm (6-8 in) deep and .92-1.2 m (3-4 ft) wide and seemed to prefer grasses, *Juncus sp.* and other green vegetation for attachment sites especially in stream sections where a constriction increased the velocity to .31-.92 m/sec (1-3 ft/sec) (Stone and Snoddy, 1969). Stone and Snoddy (1969) report that *S. dixiense* is more commonly collected with *S. slossonae* than *S. jonesi*, apparently preferring smaller cooler streams than *S. jonesi* prefers. Tarshis and Adkins (1971) report successfully transporting *S. dixiense* and other black fly larvae in aerated water in coffee cans in a styrofoam chest for distances up to 200 miles.

Habits. Nothing is reported in the literature on the adult habits.

Florida observations.

Stream Parameters

	Width	Depth	pH	Temperature		Velocity	
Mean:	7.1 m	52 cm	4.25	19°C	(66.2°F)	.67 m/sec	(2.2 ft/sec)
Min:	.6	8	3.75	13.6	(56.5)	.31	(1)
Max:	15.3	100^{+}	4.55	25	(77)	1.22	(4)

Simulium dixiense was collected from seven locations in four west Florida counties (Fig. 44). The solid dots indicate sites where well-developed, large larvae and/or pupae of *S. dixiense* were found. The open circles designate collection sites where small larvae displaying

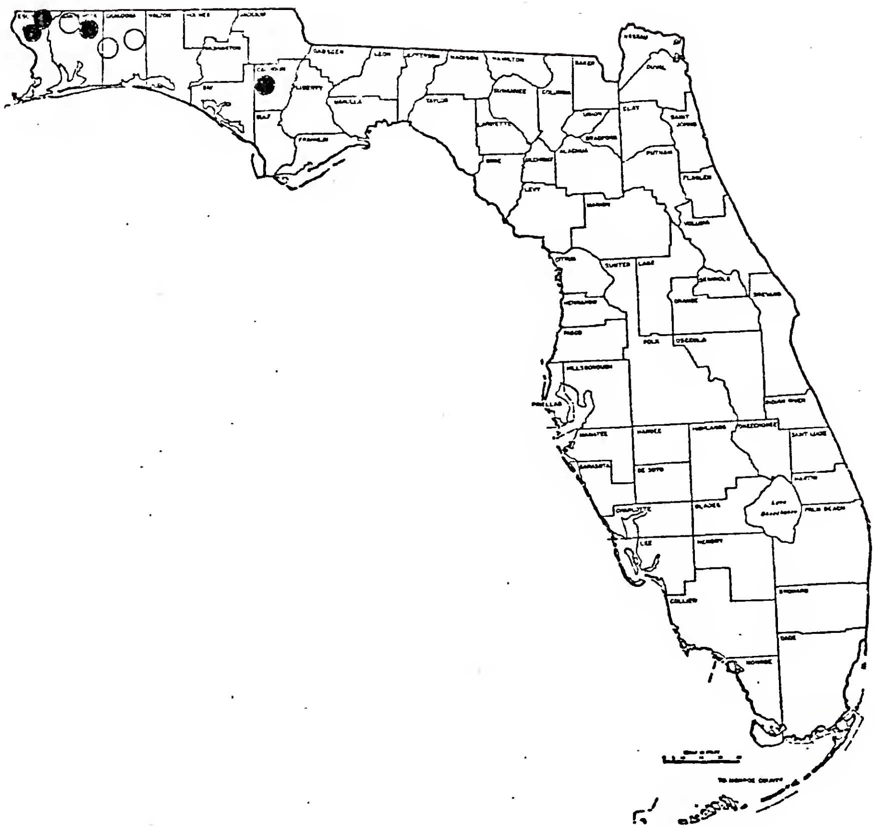

Figure 44. Collection locations for *S. dixiense* in Florida.

Figure 45. Site 74, Pine Barrens Creek, a stream inhabited by *S. dixiense*.

S. dixiense characters were recovered and the presence of this species should be further supported by collections of later immature stages. Large larvae and pupae have been collected from January through December indicating year round breeding for this species and multiple generations. *Simulium dixiense* was typically found in streams 7-15 m wide and .5 m to 1.0 m deep or deeper in sections. Immatures were recovered from the surface of the flow to about 70 cm deep. The streams exhibited a low pH reaction usually 4.0-4.5 and immatures were only collected in temperatures from 13.6°C (56.6°F) to 25°C (77°F). The stream velocity was moderately swift, usually about .46-1 m/sec (1.5-3.3 ft/sec). A typical collection site is Pine Barrens Creek (Site 74) in Escambia County (Fig. 45). Larvae and pupae were most commonly found on clean eel grass but also were collected from eel grass covered with a thin brown sediment layer or a gelatinous layer of green algae. Immatures were also found attached to thin and wide trailing bank grass, dead tree leaves, small sedges, and long thin green reeds, triangular in cross section.

Simulium dixiense was associated with five other black fly species: most frequently with *S. jonesi* but also with *S. tuberosum*, *S. verecundum*, *S. slossonae*, and *S. haysi*. Mermithid nematodes were observed on two occasions during October in three small larvae and during late December in two small larvae, in all cases at Pine Barrens Creek, Site 74.

Florida collection records for *S. dixiense*.

Calhoun Co. 49) 1975: 27 March (S,M,L,P), 6 Sept (P).

Escambia Co. 73) 1974: 18 March (S,M,P), 16 June (P). 74) 1974: 16 June (S,M,L,P), 7 Aug (S,M,P,C), 14 Oct (S,M,L,P); 1975: 19 Jan (S,L,P,A), 28 March (S,M,L,P,C,A), 12 June (S,M,L,P,C,A), 7 Sept

(S,M,L,P,C,A), 31 Dec (S,M,L); 1976: 18 April (S,M,L,P,A).

Okaloosa Co. 166) 1975: 28 March (S), 6 Sept (S), 31 Dec (S). 168) 1975: 29 March (S).

Santa Rosa Co. 195) 1974: 18 March (S,M), 7 Aug (S,M), 14 Oct (L,P); 1975: 28 March (S,M,P,A), 7 Sept (P,C), 31 Dec (P). 196) 1974: 18 March (S); 1975: 31 Dec (S).

Simulium (Phosterodoros) haysi Stone and Snoddy

Simulium haysi Stone and Snoddy, 1969, Auburn Univ. Agr. Exp. Sta. Bull. 390: 36 (female, male, pupa, larva).

Taxonomy. The holotype of this species is a female from Burnt Corn Creek, Brewton, Escambia Co., Alabama. The holotype female with pupal exuvium is deposited in the U.S. National Museum (Stone and Snoddy, 1969).

Description. The larva is 4.1 mm long. The head capsule and abdomen in a preserved state are light brown in color. The head spots are slightly darker brown and typically arranged (Fig. 46). The gular notch is broad, widest about midway along its length, extends just over half the distance to the teeth of the submentum and is pointed apically (Fig. 47). The antennae extend beyond the length of the fan stalks. The anal gills are arborescent. The ventral tubercles are large.

The pupa is 2.5-3 mm long. The head bears at least two large simple trichomes. The respiratory organ consists of 7 filaments, 6 of which arise in pairs from the basal 7th filament (Fig. 48).

The wings of the male are 1.75-2 mm long. The male is black in appearance with two silvery, iridescent anterior spots on the scutum

Figure 46. Cephalic apotome of a *S. haysi* larva.

Figure 47. Gular notch of a *S. haysi* larva.

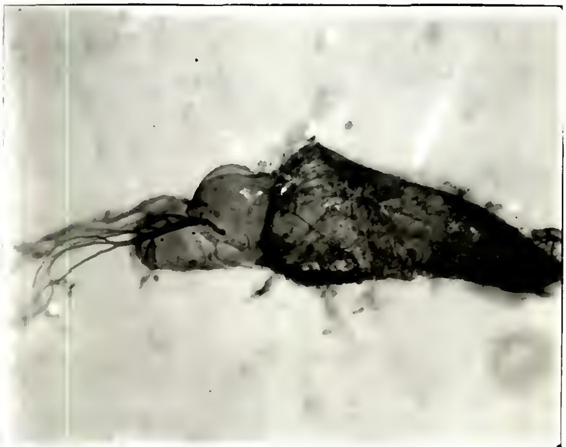

Figure 48. Pupal exuvium and cocoon of *S. haysi*.

which diverge anteriorly and posteriorly. The abdomen is black with silvery patches on the side. Stone and Snoddy (1969) indicate the terminalia are as in *S. jonesi*.

The female is small and black in appearance. The wings are almost 2.0 mm long. The frons in shining black. The clypeus is longer than wide. The scutum is dark, shiny black without pollinose anterior spots. The tibia bear shiny white patches. The genital fork has a thin stem and thin arms which branch out almost perpendicular from the stem.

Distribution. This species has been reported from a few locations in Alabama (Stone and Snoddy, 1969).

Life history. In Alabama *S. haysi* overwinters in the egg stage, is common from June to September, and completes three or more generations per year (Stone and Snoddy, 1969).

Ecology. Immatures of *S. haysi* have been collected from sedges and grasses in swift flowing, shallow sections of large streams of the Lower Coastal Plain of Alabama.

Habits. Nothing is reported on the habits of the adults in the literature.

Florida observations.

Stream Parameters

	Width	Depth	pH	Temperature		Velocity	
Mean:	12.4 m	52.8 cm	4.1	21.1°C	(70.1°F)	1 m/sec	(3.3 ft/sec)
Min:	5	15	3.75	17.2	(63)	.436	(1.43)
Max:	15.25	100	4.4	22.8	(73)	1.38	(4.54)

Mature larvae and pupae of *S. haysi* were recovered from only one stream in Florida, Big Juniper Creek, Site 195 (Fig. 49). The stream parameters above summarize stream conditions prevalent when the immatures were

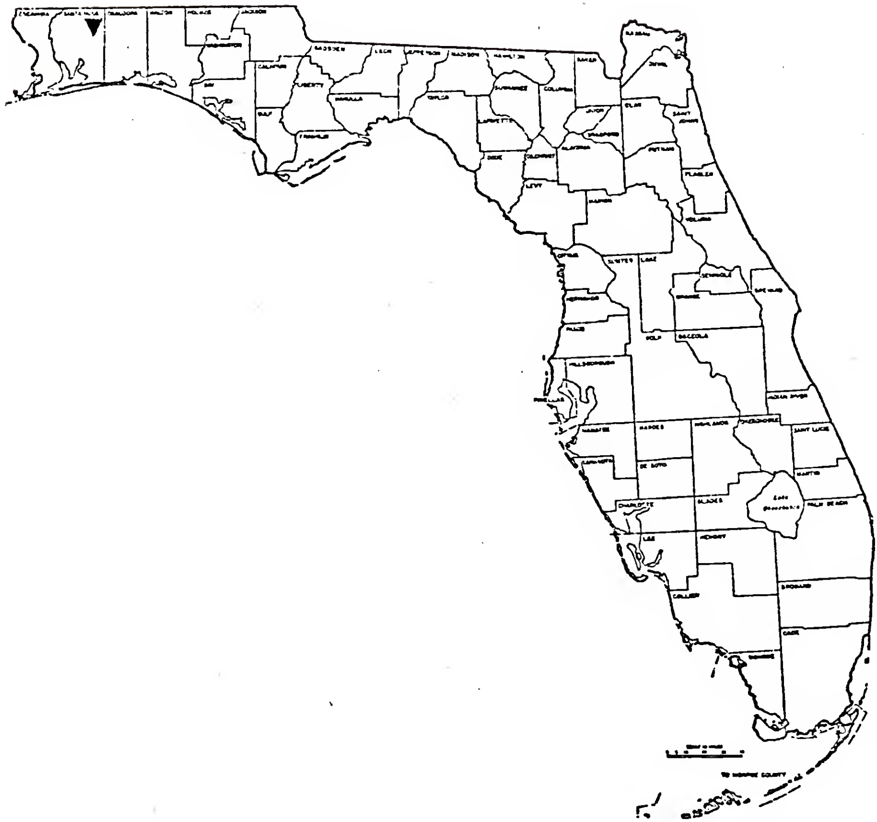

Figure 49. Location of the collection site for *S. haysi* in Florida.

Figure 50. Site 195, Juniper Creek, where *S. haysi* was collected.

collected at this site. The earliest collection occurred on 12 June and the latest on 14 October. Big Juniper Creek (Fig. 50) is a fairly large, rapidly flowing stream with a firm yellow clay or limestone substrate and large rock-like clay boulders often covered with a thick layer of slippery green algae. The creek is located in a steep-sided tree and bush-covered gorge about 20 m below the highway. The flow is shallow on the east side but on the west side a channel 1 m deep occurs. Little green vegetation occurs in the flow except some small trailing tree and bush leaves and a few sedges and clumps of grass. Immatures were found on the hard, rock-like clay and on twigs. Only small numbers of mature larvae and pupae, never more than twenty specimens per visit, were collected although some of the small unidentifiable *Phosterodoros* larvae collected at the same times may include more representatives of this species. *Simulium haysi* was associated with *S. jonesi, S. tuberosum, S. dixiense* and *S. slossonae* at this location.

At the Chipola River, Site 50, in Calhoun County, one medium-aged larvae with white histoblasts was collected in March. When mounted on a slide these histoblasts appeared to have seven filaments like the respiratory organ of *S. haysi*. Three subsequent visits to this site yielded no further *S. haysi*-like specimens.

Florida collection records for *S. haysi*.

Santa Rosa Co. 195) 1974: 16 June (L,P), 7 Aug (L,P,A), 14 Oct (L,P,A); 1975: 12 June (P), 7 Sept (P).

Simulium (Phosterodoros) jenningsi Malloch

Simulium jenningsi Malloch, 1914, U.S. Dept. Agr. Bur. Entomol. Tech. Ser. 26: 41 (female, male, larva, pupa).

Simulium nigroparvum Twinn, 1936, Can. J. Res., D, 14: 142 (female, male, pupa).

Simulium nigroparvum — Underhill, 1944, Va. Agr. Exp. Sta. Tech. Bull. 94: 1-32 (female, male, pupa, larva).

Simulium jenningsi — Davies, Peterson, and Wood, 1962, Proc. Entomol. Soc. Ontario 92: 118 (female, male, pupa).

Simulium jenningsi — Wood, Peterson, Davies, and Gyorkos, 1962, Proc. Entomol. Soc. Ontario 93: 112 (larva).

Simulium jenningsi — Stone, 1964, Conn. State Geol. and Natur. Hist. Surv. Bull. 97: 44 (female, male, larva, pupa).

Taxonomy. Malloch (1914) described a female of *S. jenningsi* as the holotype which was deposited in the U.S. National Museum. The type locality is Plummers Island, Maryland (Davies et al., 1962). Stone and Snoddy (1969) created the subgenus *Phosterodoros* to encompass the species in the former *S. jenningsi* complex which are basically similar and likely share a common ancestor, possibly the tolerant and widespread *S. jenningsi*.

Description. The larvae are about 5 mm long. The abdomen is light brown, sometimes with a reddish hue. The head spots are dark on a light yellow brown head capsule (Fig. 51). The throat cleft is broadly sagittiform, usually pointed anteriorly and extends about halfway to the submental teeth (Fig. 52). The respiratory histoblasts contain ten thin filaments.

The pupa is about 2.5 mm long. The respiratory organs each consist of ten filaments in a 2, 2, 3, 3 arrangement from dorsal to ventral. The cocoon is tightly woven, slipper-shaped and has a large aperture on each side anteriorly (Fig. 53).

Figure 51. Cephalic apotome of a *S. jenningsi* larva.

Figure 52. Gular notch of a *S. jenningsi* larva.

Figure 53. A pupa and cocoon of *S. jenningsi*.

Figure 54. *S. jenningsi* male terminalia.

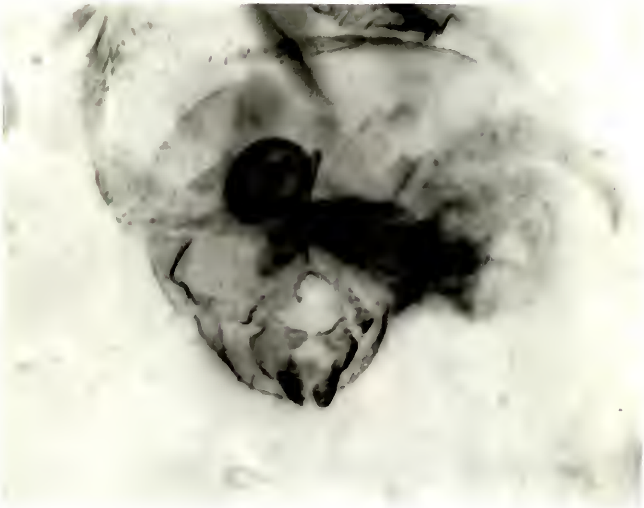

Figure 55. Genitalia of a female of *S. jenningsi*.

The male, about 2.25 mm long, is velvety black in appearance with shiny silvery areas at both the anterior lateral angles and on the posterior third of the scutum. The ventral plate in ventral view is longer than wide, in end view displays a median section roundly elongate and knobbed or pointed distally; the distimeres are three times longer than wide and tapering (Fig. 54).

The female wing is about 2.1 mm long. The female has a shiny dark brown scutum with many tiny golden appressed hairs and gray pollinosity especially laterally. The frons is shiny dark brown. The scutellum bears many long erect dark hairs posteriorly. The base of the radius and underside of the subcosta are bare. The fore coxa and femur are light yellow contrasting with the dark brown tarsal segments and distal tip of the tibia. The arms of the genital fork are fairly thin, widely separated with a slender dark dorsal projection and a more broad weakly sclerotized ventral projection (Fig. 55).

Distribution. Stone (1964) reported that *S. jenningsi* was widely distributed in eastern North America from Manitoba to Maine and south to Texas and Florida. Stone and Snoddy (1969), mentioning difficulties in separating adults in the subgenus *Phosterodoros*, restricted the distribution of *S. jenningsi* to pupal records and listed the species from Alabama, Connecticut, Kentucky, Maryland, Michigan, New York, Virginia, Wisconsin and Ontario, Canada. Crans and McCuiston (1970a) report *S. jenningsi* from New Jersey.

Life history. Underhill (1939) collected immature stages of *S. jenningsi* (as *S. nigroparvum*) in streams from April until November and suggested overwintering occurred as an egg or immature larva. Stone and Snoddy (1969) found no larvae during late winter and suggested that eggs

overwinter with hatching occurring in March or April. Immature stages of *S. jenningsi* were found in central Alabama from June through November (Stone and Snoddy, 1969). Anderson and Dicke (1960) report that in the cooler water the first generation larvae require five to six weeks to mature, indicate the pupae take five to seven days to develop and mention that the adults of the first generation in Wisconsin are usually on the wing by the third week in May. Stone and Snoddy (1969) report that five or more generations are completed each year in Alabama.

Ecology. Underhill (1944) found the immature stages of *S. jenningsi* (as *S. nigroparvum*) to be most abundant in clear streams at least 7.6-9.2 m (25 to 30 ft) wide, 28-61 cm (15-24 in) deep that flowed 1.2-1.8 m/sec (4-6 ft/sec) and contained trailing water willow *Dianthera* (now *Justicia*) *americana*. Anderson and Dicke (1960) found immatures in the clear, shallow, rocky, swift portions of young rivers 2.5-15 cm (1-6 in) below the surface on vegetation and 46 cm (18 in) deep on rocks. Stone and Snoddy (1969) mention *S. jenningsi* prefers the rapids sections of unpolluted large inland streams and rivers and immatures are found in water about 15-28°C and nearly neutral in pH.

Habits. Underhill (1944) suggested that adults may travel 32-48 km (20-30 mi) from their breeding locations. After being unable to find egg masses Underhill (1944) proposed that females deposited eggs into the streams while flying. This species has been observed in Virginia feeding on turkeys, horses, mules, cattle and, rarely, on man (Underhill, 1939, 1944). Stone (1964) mentions that it is difficult to separate *S. jenningsi* from closely related adults and suggests records of feedings on turkeys may actually be the activities of a close relative. The favorite hosts are probably cattle and horses (Stone, 1964). Stone and

Snoddy (1969) report that *S. jenningsi* is an annoying pest in the vicinity of Washington D.C. throughout the summer.

Florida observations.

Stream Parameters

	Width	Depth	pH	Temperature		Velocity	
Mean:	9.42 m	48.93 cm	6.05	21.3°C	(70.4°F)	.52 m/sec	(1.71 ft/sec)
Min:	.25	2	3.75	9.4	(49)	.15	(.5)
Max:	100	500	7.55	29.4	(85)	1.78	(5.85)

Simulium jenningsi is reported from 21 counties and 33 different locations in Florida at all times of the year with records based primarily on pupae and mature larvae with characteristic respiratory histoblasts (Fig. 56). Site 63 is marked at Biscayne Bay in Dade Co. to call attention to a record in Malloch (1914) of an adult, apparently of this species, collected by Mrs. A.T. Slosson.

Simulium jenningsi sometimes occurs in fairly wide or very wide and deep flows such as the Santa Fe River (Site 58), the Alapaha River (99), the Waccasassa River (136, 137) and the Withlacoochee (151) but often just sporadically or consistently in low numbers. Larger and more constant populations were encountered in smaller streams, generally under 5 m wide, with a fairly wide range of current characteristics: a flow to Newnan's Lake (18), Joshua Creek (65), Banana Creek (106), Blackwater Creek (113), Kettle Creek (130), Site 141 at Gulf Hammock (Fig. 57), Howell Creek (202) and Rocky Creek (213). These streams exhibited a pH approaching neutrality (about 6.5-7.5) and all contained some trailing vegetation, often grass. In other respects such as substrate (sand-mud, sand, shells, rocks, concrete) and velocity (.3-1.83 m/sec = 1-6 ft/sec) they varied considerably. A tendency for larger populations to occur

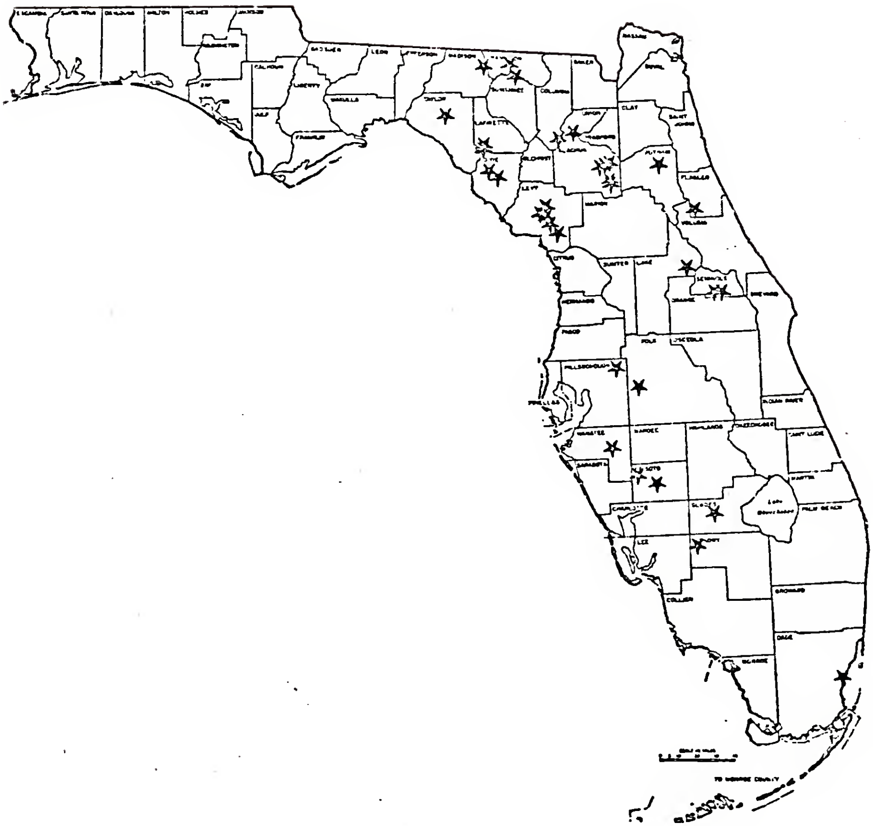

Figure 56. Collection locations for *S. jenningsi* in Florida.

Figure 57. Site 141 at Gulf Hammock where *S. jenningsi* was collected.

in faster flows, .53-.92 m/sec (1.75-3 ft/sec) was observed. Immatures were found attached to grass, eel grass, cattail blades, trailing water willow leaves, a wide variety of additional aquatic vegetation including trapped water hyacinths plus dead leaves, pine needles, shells, and rocks. *S. jenningsi* was associated with 10 other black fly species, most frequently with *S. lakei* and *S. taxodium* and much less frequently with *S. slossonae, S. tuberosum* and *S. jonesi*. This species has multiple generations each year in Florida.

It was noted that with the Florida *S. jenningsi* pupae the 5th (from the dorsal) respiratory filament consistently rises dorsally from the common base of filaments 6 and 7 while figures in the literature (Underhill, 1944 and Stone and Snoddy, 1969) illustrate a ventral origin for the single filament of the first triplet. The single filament of the 2nd (most ventral) triplet rises ventrally as typically illustrated. The surface of the respiratory filaments appears to be more smooth than the rather sharply ridged surface described for *S. jenningsi* in other areas and the filaments appear speckled like those of *S. nyssa*. Variations in the number of filaments from the normal 10 and 10 (9 and 10, 10 and 11) have been noted as has considerable difference in the point of bifurcation of the filaments. The ventral plate (Fig. 54) appears more like that illustrated for *S. jenningsi* in Davies et al. (1962) than in Stone and Snoddy (1969).

Florida collection records for *S. jenningsi*.

Alachua Co. 18) 1974: 12 Jan (P), 30 June (P), 24 July (P), 24 Aug (P), 22 Sept (P), 25 Oct (L,P), 23 Nov (P); 1975: 11 May (P), 14 June (L,P), 1 Sept (L), 17 Oct (P), 10 Dec (P). 20) 1974: 6 July (P), 29 Aug (P); 1975: 31 Jan (P), 18 April (L,P), 26 July (P), 3 Oct

(L,P,A). 21) 1974: 11 April (P); 1975: 24 Jan (L), 27 Sept (P). 22) 1974: 12 Sept (P); 1975: 29 April (P), 30 Aug (P), 17 Oct (P). 24) 1974: 25 May (L,P).

Columbia Co. 58) 1974: 21 Sept (P); 1976: 24 Jan (P).

Dade Co. 63) — A? — Mrs. A.T. Slosson.

Desoto Co. 65) 1975: 12 Sept (P,A), 22 Dec (P). 66) 1975: 22 Dec (L).

Dixie Co. 68) 1975: 26 March (L). 69) 1975: 26 March (L).

Flagler Co. 77) 1974: 25 May (P); 1975: 5 April (L,P).

Glades Co. 95) 1974: 21 March (P); 1975: 9 Aug (P).

Hamilton Co. 99) 1975: 23 June (P), 3 Aug (L,P).

Hendry Co. 106) 1974: 29 Nov (L,P); 1975: 21 March (L,P), 3 July (L,P,A), 31 May (L,P), 9 Aug (P).

Hillsborough Co. 113) 1975: 22 March (L,P), 27 May (L,P), 11 Sept (L,P,A), 21 Dec (L,P).

Lafayette Co. 129) 1974: 5 Aug (P), 12 Oct (P,A); 1975: 17 Jan (L), 26 March (P), 28 Dec (P). 130) 1974: 5 Aug (L,P), 12 Oct (P); 1975: 26 March (L,P), 23 Aug (L,P), 28 Dec (P).

Lake Co. 170) 1974: 11 July (P), 3 Sept (L).

Levy Co. 135) 1974: 3 Aug (L,P), 15 Sept (L,P); 1975: 31 Jan (P), 23 March (L,P), 1 Sept (P). 136) 1974: 18 June (P). 137) 1974: 2 March (P), 27 April (P), 3 Aug (L,P), 15 Sept (P), 2 Nov (P); 1975: 23 March (P), 23 Oct (P). 139) 1975: 30 Oct (P), 21 Dec (P,A). 141) 1975: 17 May (L,P,A), 8 July (P), 1 Sept (P), 30 Oct (P).

Madison Co. 151) 1974: 8 Aug (P); 1976: 14 Feb (P).

Manatee Co. 157) 1975: 12 Sept (P).

Polk Co. 182) 1975: 22 March (P), 21 Dec (L,P).

Putnam Co. 189) 1975: 12 Feb (L).

Seminole Co. 202) 1974: 21 March (L,P,A), 3 Sept (P); 1975: 15 March (P), 4 July (L,P), 31 Oct (P). 203) 1974: 3 Sept (P).

Suwanee Co. 208) 1974: 17 Sept (P); 1975: 24 Oct (P).

Taylor Co. 213) 1974: 14 June (L,P), 5 Aug (L,P), 12 Oct (P); 1975: 17 Jan (L,P), 26 March (L,P), 23 Aug (L,P).

Union Co. 217) 1974: 24 Aug (P), 9 Oct (P).

Simulium (Phosterodoros) jonesi Stone and Snoddy

Simulium jonesi Stone and Snoddy, 1969, Auburn Univ. Agr. Exp. Sta. Bull. 390: 29 (female, male, pupa, larva).

Taxonomy. Stone and Snoddy created a new subgenus, *Phosterodoros*, in 1969 and *S. jonesi* was one of eleven new species named at that time. Stone and Snoddy (1969) indicate that the type locality is Fish River, Baldwin Co., Alabama, and that the female holotype with its pupal exuvium and cocoon has been deposited in the U.S. National Museum.

Description. The larva is about 4.5 mm long. The cephalic apotome is yellow with brown spots (Fig. 58). The gular notch is about as wide as long, extends slightly more than halfway to the teeth of the submentum, and may be pointed anteriorly (Fig. 59). The cephalic fans possess 49 to 54 rays each. The anal gills are compound or arborescent and the anal tubercles are well developed and conical.

The pupa is 2.5 mm long. The cocoon has convex anterior edges in lateral view and a large aperture on each side anteriorly. The pupal respiratory organ consists of a stout basal filament which tapers distally and nine additional filaments which arise from the basal one (Fig. 60).

Figure 58. Cephalic apotome of a *S. jonesi* larva.

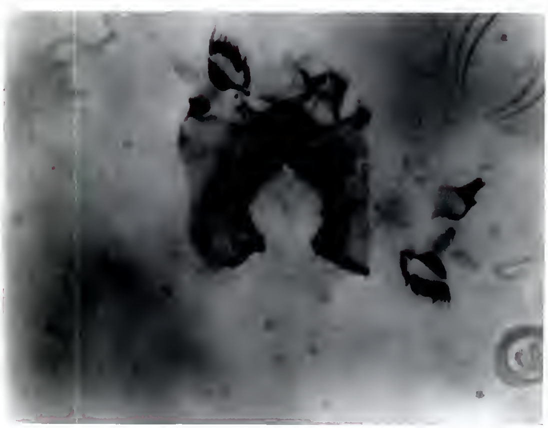

Figure 59. Gular notch of a *S. jonesi* larva.

Figure 60. Respiratory organ of a *S. jonesi* pupa.

The male is black and velvety in appearance with a pair of triangular, silvery, iridescent spots anterolaterally on the scutum. The ventral plate in ventral view has the middle portion widened distally and the arms diverge and bear posterior projections. In end view the plate is stout and tapers to a point (Fig. 61).

The female has a shiny blackish gray scutum, a shiny black frons, and shiny terminal abdominal tergites. The clypeus is about as wide as long. There are no hairs under the subcosta. The fore legs are darker brown basally on the tibia and femur and more yellowish on the distal ends of the segments. The tarsal claws lack a prominent basal projection. The genital fork is in the shape of a widespread-Y (Fig. 62).

Distribution. Stone and Snoddy (1969) mention that early collections of the species now known as *S. jonesi* were made by C.M. Jones in 1954 in Florida and South Carolina and, in addition, they list collection records in Alabama for this species.

Life history. In Alabama summer eggs are oviposited on grasses and other aquatic vegetation and the egg is believed to be the overwintering stage (Stone and Snoddy, 1969). Stone and Snoddy (1969) indicate egg hatch occurs in late February or March with mature pupae appearing by April and they state at least four generations are completed each year.

Habits. Little is recorded in the literature on the habits of *S. jonesi*. The lack of a strong basal tooth on the tarsal claw suggests this species is mammalophilic although research in Florida has revealed the females will feed on turkeys in the laboratory.

Figure 61. Terminalia of a male of *S. jonesi*.

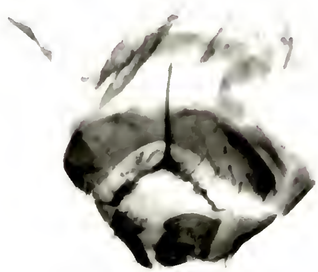

Figure 62. Genital fork and terminalia of a female of *S. jonesi*.

Florida observations.

Stream Parameters

	Width	Depth	pH	Temperature		Velocity	
Mean:	5.85 m	37.72 cm	4.7	19.6°C	(67.3°F)	.51 m/sec	(1.66 ft/sec)
Min:	.2	1	3.5	8.3	(47)	.15	(.5)
Max:	100	200^+	6.9	28.3	(83)	1.53	(5)

Simulium jonesi was found in 19 counties at 57 sites (Fig. 63). The double circles used as site designations on the map were reduced to single circles in Alachua county due to site denstiy. From Fig. 63 the restriction of this species to the southern coastal plain and northeastern Florida is apparent. It was not found south of Alachua and Putnam counties. All stages of *S. jonesi* were found year round in many streams, indicating seven or more generations per year. This species occurs in reasonable numbers in small flows 1-2 m wide such as Little Hatchet Creek (Site 18), the headwaters of Hatchet Creek (Site 36), and Site 56 and is the predominant species at specific collection sites in large flows like the Blackwater River (163) and the Bay Shoal River (221) which are 20-40 m or more wide. The streams most preferred appear to be moderately sized permanent flows about 5-8 m wide, 15-100 cm deep with trailing vegetation, trapped dead tree leaves, some shade and low pH such as South Prong Black Creek (57) and one of the paratype locations the Fenholloway River (210) (Fig. 64). Alkaline swamp flows such as more typical for *S. lakei* and its characteristic counties bordering the Gulf are generally avoided by *S. jonesi*. *Simulium jonesi* was found in streams of a more alkaline nature such as Deep Creek (191) and Blue Creek (116) but populations were usually light. In most streams *S. jonesi* immatures were collected from trailing eel grass, other grasses,

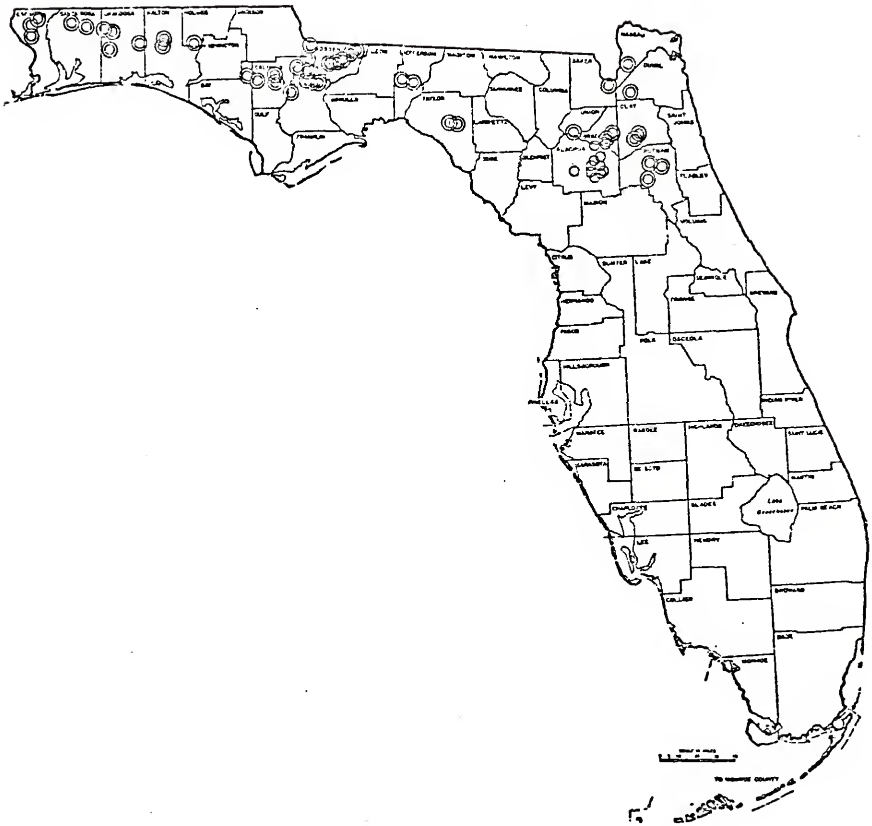

Figure 63. Florida collection locations for *S. jonesi*.

Figure 64. Site 210, the Fenholloway River, a *S. jonesi* collection location.

sedges, and other green aquatic vegetation. However in the larger sandy rivers and also the smaller sandy creeks where *S. jonesi* occurred like Piney Woods Creek (166) larvae and pupae were found most often on trapped dead tree leaves, pine needles, and twigs and limbs pressed by the flow against bridge supports. Hatchet Creek (17), before a major construction project took place in the vicinity, supported heavy populations of *S. jonesi* in a narrow (1-2 m), rapidly flowing (.76 m/sec = 2.5 ft/sec), partly shaded section with abundant eel grass and large leafed aquatic vegetation. After this habitat was destroyed the major sampling location was moved upstream about 100 meters into a deciduous and pine woods through which the creek flowed. In this shaded location the stream was a few meters wider, somewhat more shallow and was almost bare of green vegetation in the flow. However immature populations of *S. jonesi* well over 1000/.09 sq m (1 sq ft) of substrate especially during March, April, and August covered twigs, logs, dead tree leaves and pine needles. *Simulium jonesi* also occurred year round in the torrential Juniper Creek (195) where the current was usually .92-1.2 m/sec (3-4 ft/sec) and *S. jonesi* was found on small trailing water oak leaves and branches, dead leaves, pine needles and some grass.

Simulium jonesi was associated with 15 species of black flies in the streams and rivers of Florida tying *S. tuberosum* for the most associations. The most frequently associated species was *S. tuberosum*, which occurred with *S. jonesi* in sandy, limited vegetation sites and other locations on 253 occasions, and *S. slossonae* which was regularly collected (107 times) with *S. jonesi* where stream vegetation was abundant (Table 3). *Simulium verecundum* was collected with *S. jonesi*, on 44 occasions.

In more than a dozen sites from the panhandle to northeast Florida

S. jonesi larvae were found to be infected with mermithid nematodes. In the majority of instances infections were noted in less than 10% of the larvae collected at any time of the year. At South Prong Black Creek (Site 157), a good collection site for *S. jonesi*, a high incidence of mermithids was noted. In five visits infections of small larvae ranged from 11.5% (13 or 113) to 19% (7 of 36) to 26% (6 or 23) and in November 45 of 85 small larvae (53%) were found infected and no medium or large-sized larvae were recovered although a few pupae were located. Larvae infected with tiny white spheres or large white masses in the abdomen, believed to be protozoan parasites, were noted at about a dozen locations but the rates of infection were low (2-4%).

In older larvae of *S. jonesi* the anterior medial head spots were often observed to be weakly developed or absent. This characterisitc was only occasionally noted in other members of the *Phosterodoros* group where the anterior head spots were normally dark and distinct. Forms believed to be variations of *S. jonesi* were collected in Rock Creek at Torreya State Park (Site 148) and in the Apalachicola River (145). At the first location *Phosterodoros* pupae were found with respiratory filaments rising from a thicker basal one similar to the respiratory organ of *S. jonesi*; however there were only 9 filaments total and filaments 5 and 6 (numbered proximally to distally) branched off a noticeable petiole which in some specimens was at least one-third the length of the filaments themselves. At the Apalachicola River site typical *S. jonesi* pupae were mixed with other less typical pupae with respiratory organs with 10 filaments. These unusual pupae had a thick and elongate basal filament, but had filaments 5, 6 and 7 rising in a group at the end of a long petiole sometimes equal to the lengths of the filaments themselves.

Petioles which are short or nonexistent in this area of the respiratory organ are more typical of *S. jonesi*.

Simpson et al. (1956) reported finding *Simulium* species No. 58 (=*S. jonesi*, Stone and Snoddy, 1969) with other black flies in streams in Florida near two outbreaks of *L. smithi* in turkeys. During the current research immatures were collected from the Santa Fe River (Site 19) where a large population of *S. jonesi* occurred in October 1975 and of the reared females one fed on 24 October on a turkey infected with *L. smithi* in the laboratory and again fed on 27 and 28 October on two clean turkeys but no disease transmissions resulted.

Florida collection records for *S. jonesi*.

Alachua Co. 1) 1973: 2 Nov (S,M,L,P), 14 Nov (S,M,L,P), 7 Dec (S,M,P), 13 Dec (S,M,L,P); 1974: 2 Feb (S,M,L,P), 7 March (S,M,L,P), 12 April (S,M,L,P), 5 May (P), 6 July (S), 20 July (S,M,P), 29 Aug (S,M,L), 28 Sept (M,L,P), 30 Oct (S,M,L,P), 23 Nov (S,M,P,A); 1975: 10 Jan (S), 12 Feb (S,M,L,P,A), 4 April (S,M,P), 10 May (C), 21 June (S,M,L,P,A), 15 July (S,M,P), 25 Sept (S), 9 Nov (S,M,L,P,C); 1976: 21 Jan (S,M,L,P,A). 2) 1973: 11 Oct (P); 1974: 30 June (P), 17 Aug (M,P). 6) 1973: 22 Sept (S,M,L,P,A), 16 Oct (S,M,P), 25 Oct (S,M,P); 1974: 2 Feb (S,M,L,P), 7 March (S,M,P), 2 April (S,P), 18 May (S,M,P), 30 June (S,M,L,P), 30 July (S), 14 Sept (S,M,L,P,C); 1975: 15 Jan (S), 18 May (S,M), 16 Aug (S), 8 Oct (S,M,L), 6 Dec (S,M,P,C). 7) 1973: 13 Dec (S,P,A); 1974: 2 Feb (S,M,L), 14 Sept (S,P,C), 19 Oct (P), 16 Nov (S,M,L,P). 17) 1970: 17 Dec (P — Anthony); 1971: 7 Jan (P — Anthony), 21 Feb (A — Anthony); 1974: 2 Jan (S,M,L,P), 2 Feb (S,M,L,P,C), 7 March (S,M,L,P), 12 April (S,M,L,P,C,A), 5 May (S,M,L,P,C), 6 June (S,M,L,P,C), 1 July

(S,M,L,P,C), 24 Aug (S,M,L,P,C,A), 28 Sept (S,M,L,P), 4 Dec (C); 1975: 11 Jan (S,M,L,P,C), 1 March (S,M,L,P,C), 30 April (S,M,L,P,C), 21 June (S,M,L,P,C), 16 Aug (S,M,L,P,A), 8 Oct (S,M,L,P,A), 6 Dec (S,M,L,P,C). 18) 1974: 12 Jan (L,P), 2 March (P), 11 April (L), 30 June (P), 24 July (P), 24 Aug (L,P), 22 Sept (L,P), 25 Oct (L,P), 23 Nov (L,P); 1975: 10 Jan (L,P), 21 Jan (P), 11 May (L,P), 14 June (L), 10 Dec (L,P). 19) 1975: 17 July (S,M,P), 22 Oct (S,M,L,P,C,A). 19a) (L,P — D.W. Anthony). 20) 1974: 12 Jan (P), 2 March (M,L,P), 12 April (L,P), 6 July (L,P), 29 Aug (M,L,P,A), 25 Oct (M,P,A); 1975: 15 Jan (M,L,P), 31 Jan (P), 18 April (L,P), 26 July (P), 3 Oct (L,P,A). 24) 1974: 9 March (L,P), 24 May (L,P); 1976: 19 Feb (L,P). 36) 1974: 6 July (S,M,P), 24 Aug (C); 1975: 15 Jan (S), 4 April (S), 22 May (S), 22 Oct (S,M,L). 39) 1974: 23 Nov (P).

Bay Co. 40) 1975: 27 March (S,P), 11 June (S,P,C), 6 Sept (S), 30 Dec (P).

Baker Co. 41) 1974: 4 Jan (S,M,L,P), 1 June (S,M,L,P,C), 17 Sept (S,M,P), 6 Nov (S,M,P,C,A); 1975: 1 Feb (S,M,L), 26 April (S,M,L,P,C), 21 June (S,M,L,P,C,A), 17 Aug (S,M,L,P,C), 14 Nov (S,M,L,P,C).

Bradford Co. 42) 1973: 6 Oct (P). 45) 1974: 21 Sept (L,P), 16 Nov (L), 22 Nov (L,P,A).

Calhoun Co. 49) 1975: 27 March (S,M,C), 11 June (S,M,L,P,C), 6 Sept (M,L,P,C), 30 Dec (S). 50) 1975: 11 June (P). 52) 1975: 19 April (S,M,P).

Clay Co. 56) 1974: 4 Jan (S,M,L,P,C), 23 Feb (S), 4 May (S,M,L,P,C), 6 June (S,M,P,C), 20 July (S,M,C), 31 Aug (S,M), 25 Oct (S,M,P), 27 Nov (S,M,L,P,C); 1975: 16 Feb (S,M), 8 April (S,M,P), 21 June (S,M,P,C,A), 2 Aug (S,M,L,P), 19 Oct (S,M,L); 1976: 31 Jan (S,M).

57) 1974: 4 Jan (S,M,L,P,C), 23 Feb (S,M,L,P,C), 4 May (S,M,L,P, C,A), 6 June (S,M,L,P,C,A), 20 July (S,M,P), 21 Aug (S,M,L,P), 25 Oct (S,M,L,P), 27 Nov (S,P,C); 1975: 16 Feb (S,M,L,P,C), 18 April (S,M,L,P,C), 21 June (S,M,L,P,C,A), 2 Aug (S,M,L,P,C), 19 Oct (S,M,L,P,C,A); 1976: 31 Jan (S,M,C).

Duval Co. 70) 1975: 12 March (L,P), 4 May (S,M,P,C), 1 July (S), 17 Aug (S,M,L,C).

Escambia Co. 73) 1974: 18 March (S,M,L), 16 June (S,M,P), 7 Aug (S,M, L,P,C), 14 Oct (S,M,L,P); 1975: 19 Jan (S), 28 March (S,M,L,P,C), 12 June (S,M,L,P,C), 7 Sept (S,M,P,C), 31 Dec (S,L,C). 74) 1974: 16 June (S,M,L,P), 7 Aug (M), 14 Oct (S,M); 1975: 19 Jan (S), 28 March (S,M,P), 12 June (S,M), 7 Sept (S), 31 Dec (S); 1976: 18 April (S,M,L,P). 75) 1974: 7 Aug (S,M,P), 14 Oct (S,M,L,P); 1975: 28 March (S), 12 June (S,M,L,P), 7 Sept (S,M,P,C), 31 Dec (S,M,L,P,C).

Gadsden Co. 81) 1974: 17 March (M,L,P). 83) 1974: 17 March (S,M,P); 1975: 18 Jan (M), 27 March (S,M), 10 June (S,L,P,A). 85) 1975: 30 Dec (S,P,C). 87) 1974: 15 June (S,M,L,P,A), 5 Aug (S,M,L,P,C), 12 Oct (S,M,P); 1975: 18 Jan (S), 27 March (S,P), 10 June (P), 5 Sept (M,P,A). 88) 1975: 30 Dec (S); 1976: 17 April (P). 90) 1974: 17 March (S,P), 6 Aug (L,L); 1975: 29 Dec (M). 91) 1975: 10 June (M), 5 Sept (S).

Holmes Co. 116) 1974: 17 March (L,P,C), 15 June (M,L,P), 6 Aug (P,C), 13 Oct (L,P,A); 1975: 19 Jan (L), 28 March (M,L,P), 11 June (L,P,A), 6 Sept (M,L,P,C,A), 30 Dec (M,L,P); 1976: 18 April (M,L,P,C).

Jefferson Co. 124) 1974: 12 Oct (S,P), 26 March (S), 10 June (S), 23 Aug (S,M), 29 Dec (S). 125) 1974: 5 Aug (S,M,L,P), 12 Oct (S,M,L,P); 1975: 17 Jan (S), 26 March (S,M,L,P), 10 June (S,M,L, P,C,A), 23 Aug (S,M,L,P,C), 29 Dec (S,M,P).

Liberty Co. 142) 1974: 15 June (S), 13 Oct (S,P,A); 1975: 27 March (S,M), 11 June (S,L,C), 24 Aug (S,M,L,P,A). 144) 1973: 18 Dec (S,M,L); 1974: 17 March (S,M,L,P,A), 15 June (S,M,L,P), 6 Aug (S,M,L,P,C), 13 Oct (S,M,L,P,A); 1975: 18 Jan (S,M,P,C), 27 March (S,M,L), 11 June (S,P,C), 24 Aug (S,M,L), 30 Dec (S,P,C). 145) 1976: 17 April (P,C). 146) 1973: 18 Dec (S); 1974: 15 June (S,M,L,P,C), 6 Aug (P,A), 13 Oct (S,M,L,P); 1975: 18 Jan (S,M,L,P,A), 27 March (S,M,L,P,C), 11 June (S), 24 Aug (S,M,L,P,C,A), 30 Dec (S,P). 147) 1975: 27 March (S). 148) 1974: 15 June (S), 13 Oct (S,L,P); 1975: 27 March (S,L,P), 11 June (S,M,L), 6 Sept (S,M,L,P).

Nassau Co. 159) 1974: 24 Aug (M); 1975: 26 Sept (M).

Okaloosa Co. 163) 1974: 18 March (S,M), 16 June (S,P,C), 7 Aug (S,M, L,P), 14 Oct (S,M,L,P,A); 1975: 19 Jan (S,M,L,P,C), 28 March (S,M), 12 June (S,M,L,P), 7 Sept (S,M,P,C), 31 Dec (S,M,P). 164) 1975: 29 March (S,M,L,P,C), 6 Sept (S). 165) 1974: 18 March (S,M,L,P,C), 16 June (S,M), 7 Aug (S,M,C), 14 Oct (S,M,P,A); 1975: 19 Jan (S), 28 March (S,M), 12 June (S,M,L), 31 Dec (S,M,C). 166) 1974: 18 March (S,M,L,P,C), 16 June (S,M,P), 7 Aug (S,M,L,P), 14 Oct (S,M,P); 1975: 19 Jan (S,M,P), 28 March (S,M,L,P,C), 12 June (S,M,L,P,C), 6 Sept (S,M,L,P,C), 31 Dec (S,M,L,P,C).

Putnam Co. 187) 1975: 25 Sept (S,P). 189) 1974: 17 Aug (P,A). 191) 1974: 19 Jan (S,M,P), 14 April (S), 17 Aug (S,M,P,C), 23 Nov (S,M,L); 1975: 5 April (S,M,P,C), 26 June (S,M,L,C), 31 Oct (S,M).

Santa Rosa Co. 195) 1974: 18 March (M,L,P), 16 June (P), 7 Aug (L,P, C,A), 14 Oct (L,P,A); 1975: 28 March (L,P), 12 June (M,L,P,A), 7 Sept (M,L,P), 31 Dec (M,P); 1976: 18 April (M,L,P). 196) 1974: 18 March (S,M,L,C), 16 June (S,M,P), 7 Aug (M,P), 14 Oct (S,M,L,P);

1975: 28 March (S), 12 June (S,L).

Taylor Co. 210) 1954: 8 April (Larvae, Pupae — C.M. Jones); 1973: 17 Dec (S,M,L,P); 1974: 16 March (S,M,L,P), 14 June (S,M,L,P), 5 Aug (S), 12 Oct (S,M,C); 1975: 17 Jan (S,M), 26 March (S,M), 10 June (S,M,L,P), 23 Aug (S), 28 Dec (S). 211) 1975: 17 Jan (L).

Union Co. 217) 1974: 5 Jan (M), 24 Aug (L,P,A), 9 Oct (L,P); 1975: 1 June (P), 22 July (L).

Walton Co. 220) 1974: 18 March (S,M), 16 June (S,M,L,P), 7 Aug (S,M,L,P), 13 Oct (S,M,L,P,A); 1975: 19 Jan (S,P), 28 March (S,P,C), 11 June (S,M,L,P,C), 6 Sept (S,M,P,C), 21 Dec (S,P,C). 221) 1974: 18 March (S,M), 16 June (S,M,L,P), 7 Aug (M,C), 13 Oct (S,M,L,P,A); 1975: 19 Jan (S,M,P), 28 March (S,M,L,P,C,A), 11 June (S,M,L,P,C,A), 6 Sept (S,M,L,P,C), 31 Dec (L,P,C). 223) 1975: 28 March (S,M,L,P,A), 11 June (S,M,L,P), 6 Sept (S,M,L,C).

Simulium (Phosterodoros) lakei Snoddy

Simulium lakei Snoddy, 1976, J. Georgia Entomol. Soc. 11(2): 173 (larva, pupa, female, male).

Taxonomy. Snoddy (1976) described and named *S. lakei* which is the fourteenth species to be designated in the *Phosterodoros* group. The holotype male was collected from Blackbird Creek, Blackbird State Forest, Delaware and it with its associated pupal exuvium and cocoon is located in the U.S. National Museum.

Description. The older larvae are 5-5.5 mm long. Preserved specimens are brown-gray in color with brown head spots on a yellow-brown head capsule. The pattern of the spots on the cephalic apotome

is as in Fig. 65. The distal portion of the antenna extends just beyond the cephalic fan stalk. The gular notch is broadly saggittiform (Fig. 66) with faint lateral spots bordering the notch. The cephalic fans contain 57-60 rays. The anal tubercles are conspicuous and the anal gills are arborescent.

The pupa is about 3.5 mm long. The anterior lateral openings of the cocoon are usually large. The respiratory filaments number 9 with filaments 1-6 arising in pairs off short petioles and filaments 7-9 (most ventral) rising off a longer base (Fig. 67).

The wings of the male are about 2 mm long. The scape and pedicel of the antenna are reddish brown and lighter in color than the black flagellum. The scutum is black and velvety with bright anterior lateral patches (Fig. 68). The abdomen is also black and velvety with iridescent spots on the side of segments 2, 6, and 7. The middle portion of the ventral plate expands distally and the ventral plate and the distimeres appear as in Fig. 69.

The wings of the female are about 2 mm long. The frons is shiny brownish black. The width of the clypeus is about equal to its length. There are no hairs under the subcosta. The scutum is shiny grayish black. The tarsi are light yellow brown. The genital fork appears as in Fig. 70.

Distribution. Snoddy (1976), in addition to the holotype location in Delaware, records *S. lakei* from South Carolina and states this species may extend into western Georgia although it was not found in collections from West Central Georgia.

Life history, ecology, habits. Little else is recorded in the literature concerning the biology and ecology of *S. lakei*. Larvae,

Figure 65. Cephalic apotome of a *S. lakei* larva.

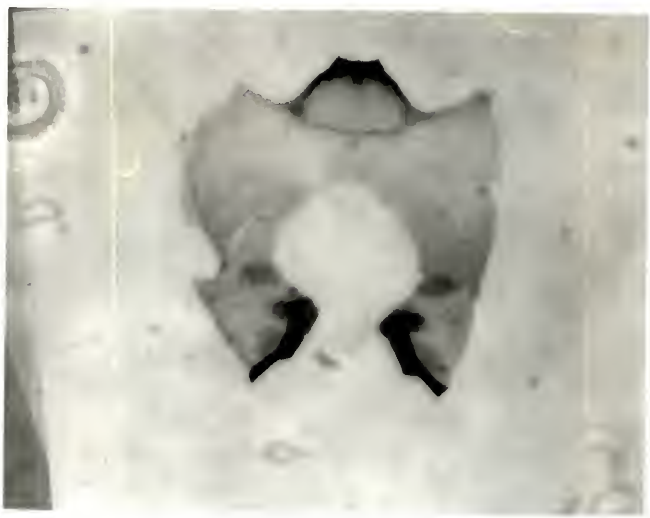

Figure 66. Gular notch and hypostomium of a *S. lakei* larva.

Figure 67. Pupa and cocoon of *S. lakei*.

Figure 68. Dorsal view of a male of *S. lakei*.

Figure 69. Terminalia of a male of *S. lakei*.

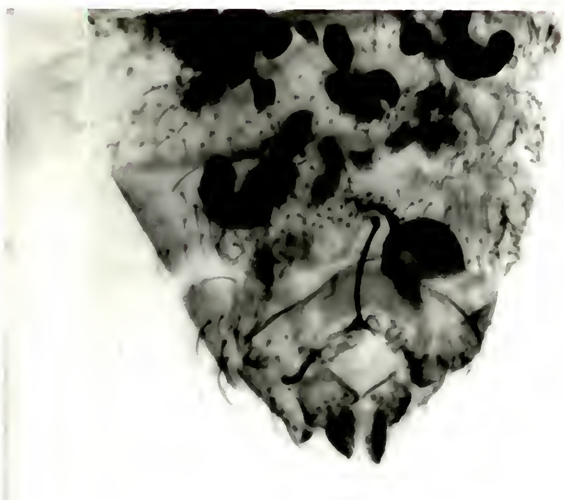

Figure 70. Terminalia of a female of *S. lakei*.

pupae and adults (reared) were obtained in Delaware from early May to late October and larvae and pupae were found in South Carolina from late March through August (Snoddy,1976).

Florida observations.

Stream Parameters

	Width	Depth	pH	Temperature		Velocity	
Mean:	9.93 m	54.61 cm	6.01	20.8°C	(69.4°F)	.49 m/sec	(1.62 ft/sec)
Min:	.2	1.7	3.75	7.2	(45)	.15	(.5)
Max:	100	1000	7.55	29.4	(85)	2	(6.66)

Based primarily on the similarity between the structures of Florida specimens and the description published in Snoddy (1976) for the pupal respiratory organ and confirmation of determinations by Dr. E.L. Snoddy of submitted Florida specimens this species has been called *S. lakei*, with the observed morphological variations listed below. Specimens have been collected from 43 locations in 24 counties (Fig. 71). This species has not been positively identified from collections in Florida west of Jefferson County but occurs widely in northeastern Florida and ranges south to west of Lake Okeechobee. This species is mulivoltine and in permanent streams larvae and pupae may be found all year long. *S. lakei* occurs in small temporary flows such as Otter Creek (Site 135, Fig. 72), Rutland Creek (Site 206) and Rocky Creek (213); smallish permanent flows such as Joshua Creek (65), a flow to Newnan's Lake (18), Site 141, and Howell Creek (202); and in larger permanent flows such as the Santa Fe River at Oleno State Park (58), Blackwater Creek in Hillsborough Co. (113), the Waccasassa River (136, 137) and the Withlacoochee River (151). At least some of these flows are spring fed being characteristically near neutrality or just below in pH reaction and all are

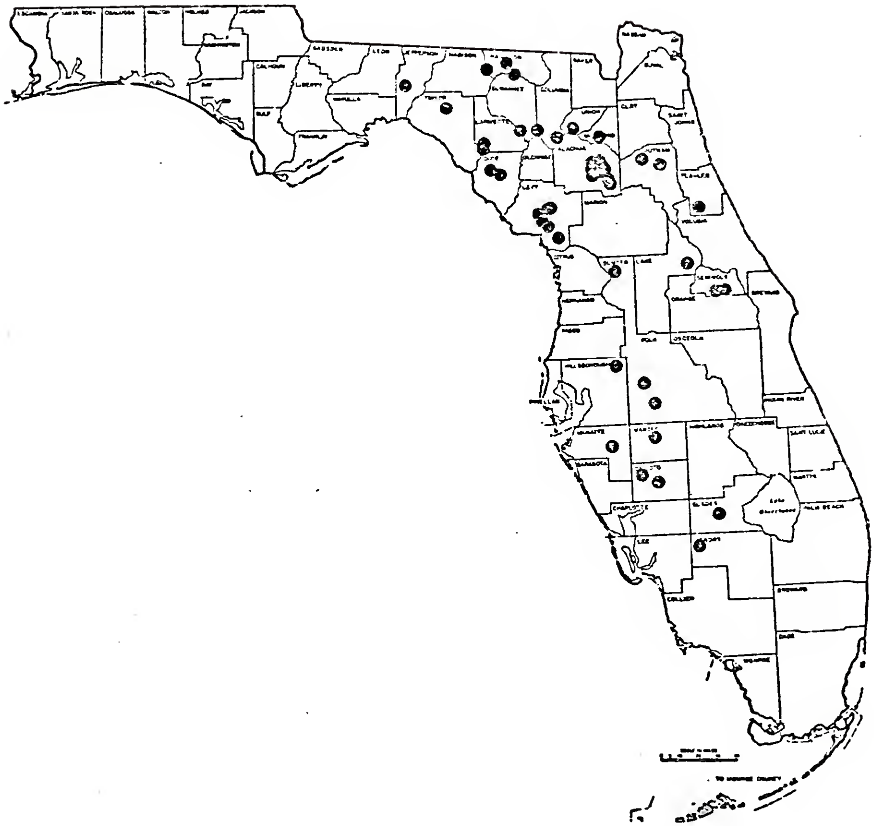

Figure 71. Collection locations for *S. lakei* in Florida.

Figure 72. Site 135, Otter Creek, a collection site for *S. lakei*.

moderately to very swift flowing with currents at constrictions formed by boulders in the Santa Fe River and flows pouring off the concrete at Howell Creek reaching 1.2-1.8 m/sec (4-6 ft/sec) or more. Other collection sites such as Fisheating Creek (95), Blackwater Creek in Lake Co. (170) and Rice Creek (189) were slower flowing and yielded smaller populations of *S. lakei*. This species was also found to occur year round in Little Haw Creek (77) which was fairly deep and flowed about .46 m/sec (1.5 ft/sec) and Lochloosa Creek (21) which was fairly shallow and slow flowing (.3-.46 m/sec = 1-1.5 ft/sec); both of these streams were more acidic than is typical for the habitats of this species and averaged around 4.5 pH reaction. The substrate for most sites was sand with some pebbles, rocks, or boulders occurring in the flow. Immatures were typically found attached to a wide range of green aquatic, trailing and trapped vegetation including grasses, sedges, leaves of cattail, smartweed, pickerelweed, Hemlock and water willow, twigs, dead tree leaves, pine needles and algae or moss on rocks. On eel grass pupae were found often situated on the middle of the blade parallel to the long axis and facing the blade tip. On willow and other tree leaves pupae frequently attached near the outer leaf edge near the serrations parallel with the main rib and were found on both sides of the leaves. The largest populations of immatures were encountered usually during the cooler part of the year from November through April; however, heavy populations were noticed in some permanent flows at other times such as during August in Kettle Creek (Site 130).

Simulium lakei was found in Florida streams and rivers at least once with nine other black fly species (Table 3). Most frequently *S. lakei* was associated with the other *Phosterodoros* species, *S. taxodium*

(150 associations) and *S. jenningsi* (92 associations). *Simulium slossonae* and *S. tuberosum* were next most commonly found with *S. lakei.*

In September 1975 a reared *S. lakei* fed on a turkey in the laboratory. A number of *S. Phosterodoros* species females which are difficult to separate without associated pupae and may have included females of *S. lakei* were captured in Manitoba traps at Fisheating Creek and elsewhere and fed on turkeys in the laboratory. However since this species lacks the tooth on the tarsal claw typical of ornithophilic species, it probably prefers mammals such as the scrub cattle which ranged near many of the breeding sites like Lochloosa Creek (22).

Morphological variations have been observed in Florida specimens of *S. lakei* compared to specimens illustrated from other areas in Snoddy (1976). Some of the Florida larvae display a gular notch which is wider and less elongate than pictured. There appear to be about 76 rows of anal hooks on the larva rather than 56-58 rows. The dorsal most respiratory filament on a number of *S. lakei* pupae, locally, rises vertically and abruptly proceeds anteriorly at a much sharper angle than the gentle curve drawn. The most ventral filament rather than separating from the common stem of filaments 7 and 8 far out from the base rises very close to the base of the petiole of 7 and 8 and the main organ stem. Many pupae have been collected with 8 filaments on one side and 9 on the other or with a 9 and 10 filament variation. The female and male terminalia generally agree with figures for *S. lakei.*

<u>Florida collection records for *S. lakei*</u>

Alachua Co. <u>1</u>) 1974: 12 April (L,P). <u>7</u>) 1974: 2 Feb (P). <u>17</u>) 1975: 16 Aug (L). <u>18</u>) 1974: 12 Jan (L,P), 2 March (L,P), 11 April (L,P), 25 May (L,P), 30 June (P), 24 July (L,P), 24 Aug (L,P), 22 Sept

(L,P), 25 Oct (L,P), 23 Nov (L,P); 1975: 10 Jan (L,P), 31 Jan (P), 8 April (L,P), 11 May (L,P,A), 14 June (P), 17 July (P), 1 Sept (L,P,A), 17 Oct (L,P,A), 10 Dec (L,P,A). 20) 1974: 12 Jan (P), 2 March (L,P), 6 July (P), 29 Aug (L,P,A), 23 Nov (P); 1975: 15 Jan (L,P), 31 Jan (L,P), 18 April (L,P), 26 July (L,P), 3 Oct (L,P). 21) 1974: 2 March (L,P), 11 April (L,P), 30 July (P), 14 Sept (L,P), 30 Oct (P); 1975: 24 Jan (L,P), 4 April (L,P), 27 Sept (L,P); 1976: 21 Jan (P), 29 Jan (P). 22) 1974: 15 Aug (L,P), 28 Aug (L,P), 12 Sept (P), 7 Dec (L,P); 1975: 24 Jan (L,P,A), 4 April (L,P), 29 April (L,P), 5 Aug (L,P), 30 Aug (L,P), 17 Oct (L,P,A); 1976: 6 March (L,P). 24) 1974: 9 March (P), 25 May (L,P); 1975: 18 May (P); 1976: 19 Feb (L,P). 39) 1974: 25 Oct (P), 23 Nov (P); 1975: 26 July (P).

Bradford Co. 45) 1974: 4 May (P), 21 Sept (P), 16 Nov (L); 1975: 22 Nov (P,A).

Columbia Co. 58) 1974: 23 Feb (P), 27 April (P), 21 Sept (L,P), 6 Nov (L,P); 1975: 12 Jan (L,P), 26 April (L,P), 21 June (P), 17 Aug (L,P), 22 Oct (L). 59) 1974: 27 April (P), 6 Nov (P).

Desoto Co. 65) 1974: 29 Nov (P); 1975: 22 March (L,P,A), 12 Sept (L,P), 22 Dec (L,P). 66) 1975: 22 Dec (P).

Dixie Co. 68) 1975: 26 March (P), 25 Aug (S,P). 69) 1975: 26 March (P).

Flagler Co. 77) 1974: 26 Jan (L,P), 25 May (L,P), 31 Aug (L,P), 7 Dec (L,P); 1975: 5 April (L,P), 9 July (L,P), 31 Oct (P).

Glades Co. 95) 1975: 28 July (P), 9 Aug (L,P).

Hamilton Co. 99) 1974: 8 Aug (L,P); 1975: 1 Feb (L), 26 April (L,P), 23 June (P), 3 Aug (L,P), 24 Oct (P).

Hardee Co. 104) 1975: 21 Dec (P).

Hendry Co. 106) 1974: 29 Nov (L,P,C); 1975: 31 May (L,P), 3 July (M,L,P), 9 Aug (P), 16 Oct (L).

Hillsborough Co. 113) 1975: 22 March (L,P), 27 May (L,P), 11 Sept (M,L), 21 Dec (L,P,A).

Jefferson Co. 125) 1974: 5 Aug (L).

Lafayette Co. 129) 1974: 16 March (L,P), 14 June (L,P,C), 5 Aug (L), 12 Oct (L,P,C); 1975: 17 Jan (L), 26 March (L,P), 23 Aug (L,P), 28 Dec (P). 130) 1974: 14 June (L,P), 5 Aug (L,P), 12 Oct (L,P); 1975: 17 Jan (L,P), 26 March (L,P), 10 June (L,P), 23 Aug (L,P), 28 Dec (P).

Lake Co. 170) 1974: 28 Nov (P).

Levy Co. 135) 1973: 7 Oct (F); 1974: 2 March (L,P), 27 April (L,P,C), 3 Aug (L,P), 15 Sept (L,P); 1975: 21 Jan (L,P), 23 March (L,P), 17 May (P), 1 Sept (L,P,A), 30 Oct (L,P,C), 21 Dec (L,P). 136) 1974: 2 March (L,P), 18 June (P), 3 Aug (L), 15 Sept (L,P,C), 2 Nov (L,P,A); 1975: 31 Jan (L,P), 23 March (L,P,A), 17 May (P), 30 Oct (L,P). 137) 1973: 29 Dec (L,P); 1974: 2 March (L,P), 27 April (L,P), 3 Aug (L,P), 15 Sept (L,P), 2 Nov (L,P,A); 1975: 31 Jan (L,P), 23 March (L,P,A), 5 July (L,P,C), 1 Sept (L,P), 30 Oct (L, P,A). 138) 1975: 31 Jan (L,P). 139) 1975: 23 March (P), 30 Oct (L,P), 21 Dec (L,P). 141) 1975: 23 March (L,P), 17 May (L,P), 8 July (L,P), 1 Sept (L,P,A), 30 Oct (L,P).

Madison Co. 151) 1974: 10 Nov (L,P); 1975: 1 Feb (P), 26 April (L,P), 23 June (P), 3 Aug (P); 1976: 14 Feb (P).

Manatee Co. 157) 1975: 12 Sept (L,P), 22 Dec (L,P).

Polk Co. 181) 1974: 29 Nov (L,P,A); 1975: 22 March (L,P,C), 21 Dec

(L,P,A). 182) 1975: 11 Sept (P), 21 Dec (L,P).

Putnam Co. 186) 1974: 25 May (L), 23 Nov (P). 189) 1974: 19 Jan (P), 14 April (P,C), 17 Aug (L,P,C), 6 Oct (P), 23 Nov (P); 1975: 12 Feb (L), 31 Oct (P).

Seminole Co. 202) 1974: 21 March (L,P), 12 May (P), 11 July (L,P), 3 Sept (L,P), 28 Nov (L,P,C); 1975: 15 March (L,P,C), 27 April (L,P,A), 4 July (L,P), 31 Oct (L,P). 203) 1974: 3 Sept (P), 28 Nov (L,P,C); 1975: 15 March (L,P,C), 31 Oct (L).

Sumter Co. 206) 1973: 21 Nov (M,P).

Suwanee Co. 207) 1973: 6 Nov (P). 208) 1974: 8 Aug (L), 17 Sept (P,A), 10 Nov (P); 1975: 1 Feb (L,P,A), 26 April (L,P,A), 23 June (P), 24 Oct (L,P); 1976: 24 Jan (L).

Taylor Co. 213) 1974: 14 June (P), 5 Aug (P), 12 Oct (L,P,A); 1975: 26 March (L,P), 23 Aug (L,P).

Union Co. 217) 1974: 23 Feb (L,P), 4 May (P), 6 July (L,P); 1975: 22 July (L,P,A).

Simulium (Phosterodoros) notiale Stone and Snoddy

Simulium notiale Stone and Snoddy, 1969, Auburn Univ. Agr. Exp. Sta. Bull. 390: 40 (female, male, pupa).

Taxonomy. A male was described and named as the holotype for this species. The type locality is Meadows Mill, Lee Co., Alabama. The holotype male with associated pupal exuvium and cocoon has been deposited in the U.S. National Museum (Stone and Snoddy, 1969).

Description. Stone and Snoddy (1969) did not positively identify the larvae. During the current research *S. notiale* larvae were observed

to have the following characteristics: the length of the larvae was 5-5.75 mm; the head capsule appeared yellow brown with dark brown head spots and the abdomen was greenish-brown which faded to gray in alcohol; the posterior lateral head spots were not slanted but positioned horizontally on the epicranial plate (Fig. 73); the head capsule appeared fairly wide with convex margins when viewed dorsally; the gular notch was broad, extended over half the distance to the submental teeth, was bordered by a pair of dark oval spots along its length and was either broadly rounded or more pointed anteriorly (Fig. 74); the anal tubercles were inconspicuous.

The pupa is 2.5-3 mm long. The respiratory organs each consist of 6 filaments in 3 pairs on short petioles with the filaments' length being about one-third to one-half the length of the pupa. The cocoon is joined anterioventrally (Fig. 75).

The male is velvety black with a pair of large triangular iridescent areas laterally on the forward part of the scutum. The velvety black area between the iridescent spots is broadly expanded anteriorly and expands posteriorly and meets a broad more dull, gray region at the rear of the scutum (Fig. 76). The abdomen is velvety black with iridescent patches along the side. Terminalia are as in Fig. 77.

The female bears wings about 2.5 mm long. The hairs are dark on the stem vein and costa. The frons is shiny black. The clypeus is thinly gray pollinose and slightly longer than wide. The scutum is shiny dark brown with a pair of pale gray spots anteriorly. The fore tibia bear elongate bright white patches that extend three-fourths of the length of the tibia. The anterior abdominal tergites are velvety black while the posterior four tergites are shiny black. Terminalia are as in Fig. 78.

Figure 73. Head spots of a *S. notiale* larva.

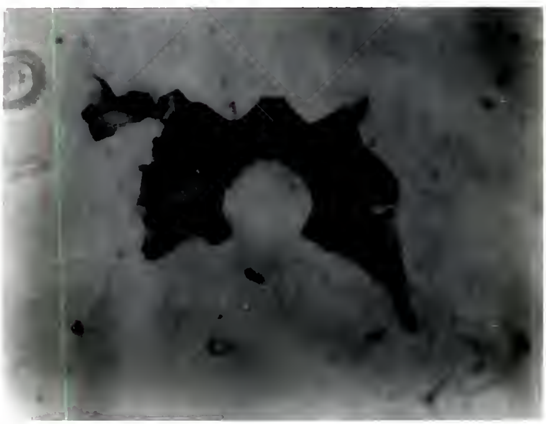

Figure 74. Gular notch of a *S. notiale* larva.

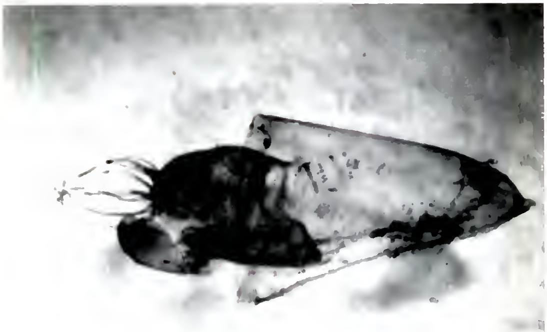

Figure 75. Pupal exuvium and cocoon of *S. notiale*.

Figure 76. Scutum of a male of *S. notiale*.

Figure 77. Terminalia of a male of *S. notiale*.

Figure 78. Terminalia of a female of *S. notiale*.

<u>Distribution</u>. *Simulium notiale* has been reported from Alabama, South Carolina and Virginia (Stone and Snoddy, 1969).

<u>Life history</u>. Stone and Snoddy (1969) found *S. notiale* only in the early spring and suggest that the eggs overwinter and that the species is univoltine.

<u>Ecology</u>. In Alabama pupae of *S. notiale* were collected in medium-sized streams with high quality water exhibiting a temperature of 13-16°C and a pH of 7.1-7.2. Pupae were found in waterfalls at the top of a dam attached to sticks in a flow of .76-.92 m/sec (2.5-3 ft/sec) where *S. venustum* also occurred.

<u>Habits</u>. The habits of this black fly are unknown.

<u>Florida observations</u>.

Stream Parameters

	Width	Depth	pH	Temperature		Velocity	
Mean:	35 m; 5.5 m	4.3 cm; 18.74 cm	4.51	18.3°C	(65°F)	1.16 m/sec	(3.79 ft/sec)
Min:	34 ; 4	3 ; 5	4.45	9.4	(49)	.67	(2.2)
Max:	36 ; 6.5	5 ; 25	4.65	25.6	(78)	1.53	(5)

Simulium notiale was found in one county, Gadsden, at two sites adjacent to each other, Sites 88 and 89, in Chattahoochee, Florida, during April, August and December (Fig. 79). All stages were present during each month; however, the smallest populations were found during December and the largest during August. In Florida this species appears multivoltine and capable of completing generations most all of the year. In the stream parameter table the width and depth figures are listed for each site separately. At Site 88 (Fig. 80) *S. notiale* immatures were collected from dead leaves, pine needles, and small amounts of grass at the crest of a rounded, concrete, power station dam about 35 m long and 6.1 m (20 ft)

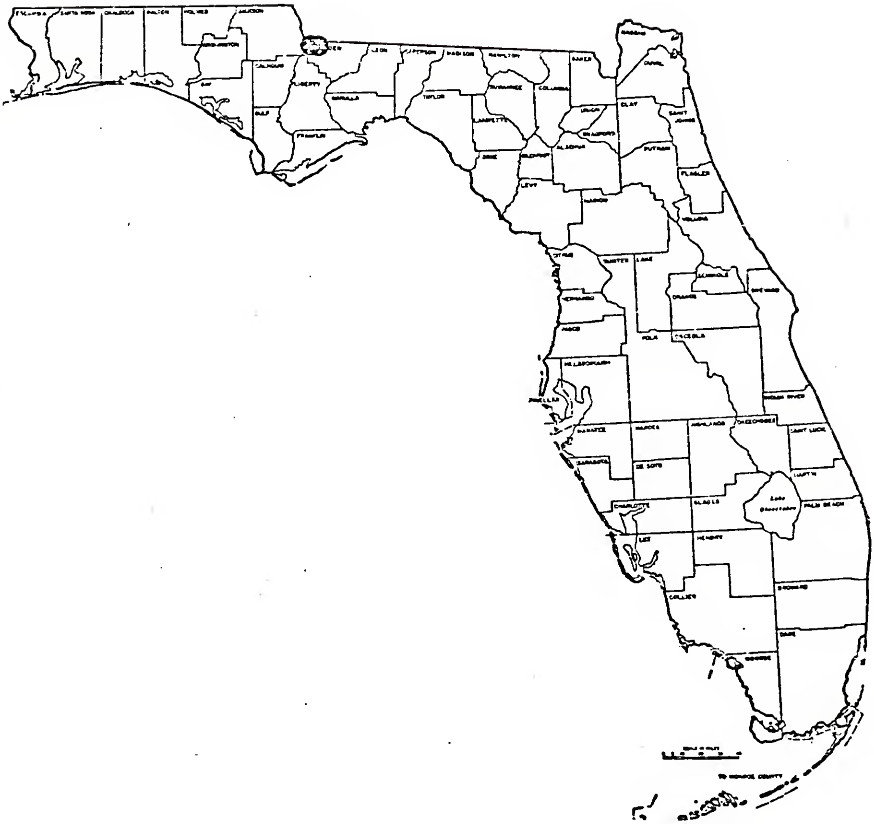

Figure 79. Collection locations for *S. notiale* in Florida.

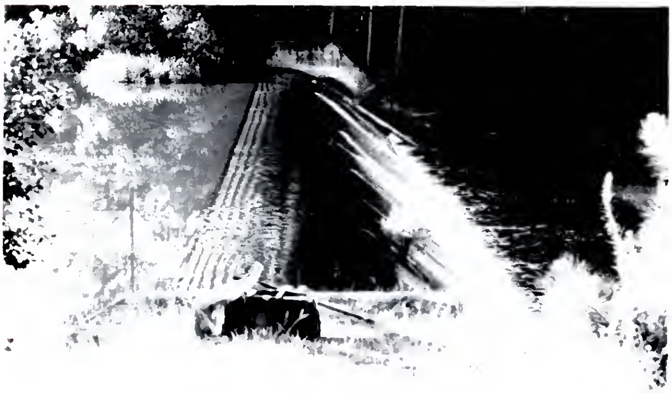

Figure 80. Site 88 at Chattahoochee where *S. notiale* immatures were found.

high. The water poured at high velocity a few centimeters deep from a large impounded lake. No *S. notiale* immatures were found attached to the concrete which was coated with a thin layer of green algae. At Site 89, Mosquito Creek, immatures were recovered a short way downstream from a second, smaller dam in a rapidly flowing, moderately deep stream about 5 m wide which flowed over exposed yellow, slippery limestone. Little vegetation occurred in the flow but black snails were abundant. Hardwoods and pines crowded in on the stream. In August large populations of *S. notiale* were found attached to outcrops of the yellow rock-like substrate, moss, pine needles, green trailing tree and bush leaves, dead leaves, and a few blades of grass. The pH of both sites was below 5.0 and the immatures were found in water from 9.4°C to 25.6°C (49-78°F). *Simulium notiale* was collected with *S. tuberosum*, *S. jonesi*, *S. decorum* and *S. verecundum* (Table 3). Two reared females each fed once on a turkey in the laboratory during August but died within two days of the feedings.

Florida collection records for *S. notiale*.

Gadsden Co. 88) 1975: 24 Aug (S,M,L,P,C), 30 Dec (S,M); 1976: 17 April (M,P). 89) 1975: 8 Aug (S,M,L,P — K. Tennessen), 24 Aug (S,M,L,P,A), 30 Dec (S,M,P,C,A); 1976: 17 April (S,M,L,P,A).

Simulium (Phosterodoros) nyssa Stone and Snoddy

Simulium nyssa Stone and Snoddy, 1969, Auburn Univ. Agr. Exp. Sta. Bull. 390: 42 (female, male, pupa).

Taxonomy. A male was described and designated the holotype of *S. nyssa* by Stone and Snoddy (1969). The type locality is Meadows Mill,

Lee Co., Alabama. The holotype male has been deposited in the U.S. National Museum.

Description. Stone and Snoddy (1969) state that the larva is apparently not separable from *S. jonesi* or *S. dixiense* without examination of the respiratory histoblast. No larvae recognizable as *S. nyssa* were collected in the current research.

The pupa is 3 mm long in a typical *Phosterodoros* cocoon with lateral apertures. The pupal respiratory organ consists of 10 thin filaments. The dorsal or posterior 4 filaments are shorter than the other filaments and arise from short petioles or are nearly sessile. The remaining 6 filaments consist of two pairs each with a long petiole and a third filament which rises off the petiole of each pair (Fig. 81).

The male has a gray pollinose clypeus, brown palpi, and a black scutum with a pair of iridescent spots near the anterior margin. The dark area between the spots widens anteriorly and posteriorly. The abdomen is deep reddish brown with the usual silvery pollinose areas (Stone and Snoddy, 1969).

The female has wings which are 1.75-2.3 mm long with dark stem vein hairs. The frons is shining black; the clypeus is lightly gray pollinose, slightly longer than wide. The scutum is shiny black with a pair of pale gray spots anteriorly and thin recumbent coppery hair. The abdomen is dark with a yellow to coppery basal fringe and shiny terminal terga (Stone and Snoddy, 1969).

Distribution. Stone and Snoddy (1969) list records for *S. nyssa* from Maine and Connecticut in the north, Oklahoma, Arkansas, and Texas out west and Virginia, Mississippi, Missouri, Kentucky, Louisiana, Alabama, South Carolina, and Florida in the south.

Figure 81. *S. nyssa* pupa and cocoon.

Life history. In Alabama four or five generations are completed each year with overwintering occurring in the egg stage (Stone and Snoddy, 1969). Sleeper (1975) reports three or four generations for *S. nyssa* in Maine.

Ecology. Stone and Snoddy (1969) report *S. nyssa* is commonly collected on river weed, *Podostemon ceratophyllum*, in shallow rapids (.76-1.06 m/sec = 2.5-3.5 ft/sec) in large streams in association with *S. underhilli* and in small streams with *S. snowi*. Immatures prefer water temperatures 13-28°C and have been collected in water with a pH reaction of 6.8-7.3. Sleeper (1975) reports that swift, cool, pure mountain streams sustain *S. nyssa* in Maine.

Habits. Stone and Snoddy (1969) observed females believed to be *S. nyssa* ovipositing on the face of a dam by flying up and down and tapping their abdomens to the trickle that slowly flowed over the dam surface. Females are reported to be primarily annoying to man which they seldom bite but are said to be serious pests of cattle, attacking the ears, stomach, genitalia and other regions. Sleeper (1975) reports *S. nyssa* is a vicious man-biter in Maine.

Florida observations.

Stream Parameters

Width	Depth	pH	Temperature	Velocity
6 m	30-90 cm	6.75	14.2°C (57.5°F)	.85 m/sec (2.8 ft/sec)

Simulium nyssa is reported from one site in each of two counties in Florida (Fig. 82). The record for Orange County refers to pupae collected in January 1947 from the Big Econlockhatchee River in Orlando. Three pupae were examined at the U.S. National Museum that were labeled

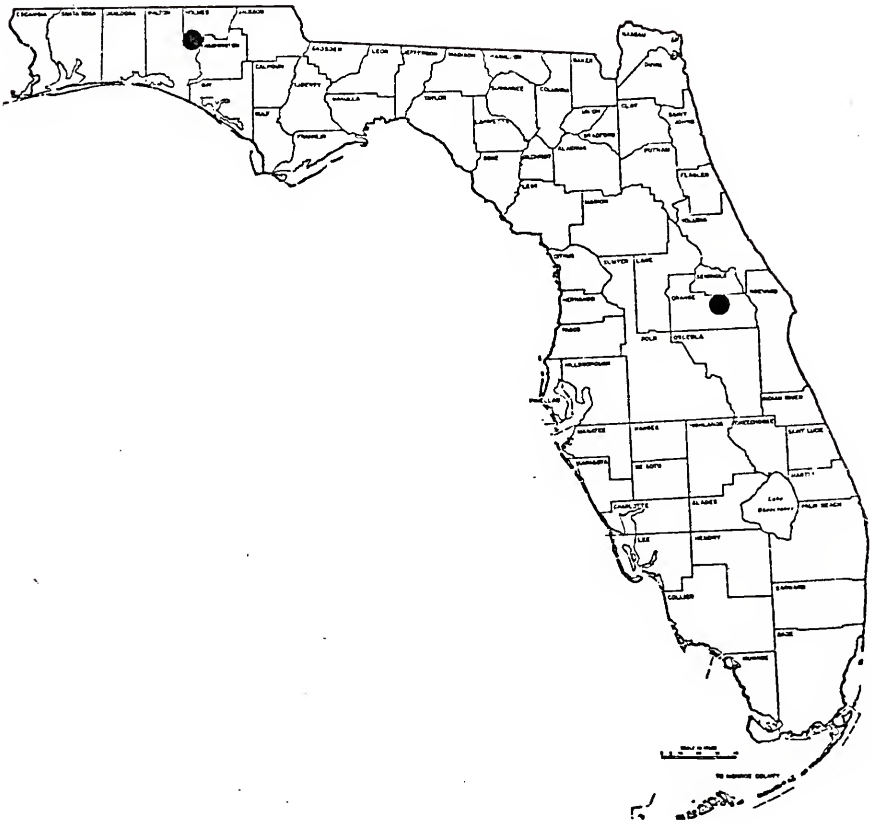

Figure 82. Collection locations for *S. nyssa* in Florida.

Figure 83. Site 116, Blue Creek, where *S. nyssa* was collected.

S. nyssa but were observed to have only 9 filaments in the respiratory organ similar to *S. lakei* which Snoddy (1976) recently described. If this is the Florida record referred to in Stone and Snoddy (1969) other specimens not examined in the current study must be on hand or the record is questionable.

In this research only one pupa which appears to be *S. nyssa* was collected, and that at Blue Creek, Site 116 (Fig. 83). The collection was made on 19 January and the pupa was attached to a thin twig. The stream parameters for Blue Creek at the time of the collection are presented above. The stream substrate consisted of planks, logs, twigs, debris and sand. Little green vegetation except trailing tree leaves occurred in the section of the stream sampled. The pupa was collected with specimens of *S. jonesi* and *S. tuberosum*. The Florida *S. nyssa* pupa has the solitary filament of each lower or anterior triplet on the respiratory organ rising ventrally off the long petioles of the other filaments instead of dorsally as figured for *S. nyssa* in Stone and Snoddy (1969). However specimens from Oklahoma observed at the National Museum and labeled *S. nyssa* appear like the Florida specimen.

Florida collection records for *S. nyssa*.

Holmes Co. 116) 1975: 19 Jan (P).

Orange Co. 171) 1947: 9 Jan (P -- H.K. Gouck).

Simulium (Phosterodoros) taxodium Snoddy and Beshear

Simulium (Simulium) taxodium Snoddy and Beshear, 1968, J. Georgia Entomol. Soc. 3(3): 123 (female, male, pupa, larva).

Taxonomy. Snoddy and Beshear (1968) designated a female with its

associated pupal exuvium and cocoon as the holotype. The type locality is Chickasawhatchee Creek at Highways 37 and 216 in Baker County, Georgia. The holotype is deposited in the U.S. National Museum. Paratypes are located in the museums of the University of Georgia at Athens and Experiment, Georgia.

Description. Fully developed larvae are 4.5 mm long. The head capsule is yellow brown with brown head spots. The abdomen of preserved specimens is yellow brown to gray brown in color. Snoddy and Beshear (1968) state the gular notch is bulbous and broadly rounded anteriorly, that each caphalic fan contains 38-42 rays and that there are 76-82 rows of anal hooks (Fig. 84 and 85).

The pupa is 2.5-3 mm long. The respiratory organs each consist of 8 filaments arranged in four pairs. The petiole of the third pair, from the dorsal, is the longest (Fig. 86). The cocoon is tightly woven, slipper-shaped and bears a pair of large anterior-lateral openings.

The wing of the male is about 2 mm long. Large bright silvery patches occur on the forward-lateral areas of the scutum and the posterior is silvery with the remainder of the scutum being velvety black. The stem vein hairs are dark brown. The abdomen is velvety black with silver patches along the side. The distimeres are elongate, taper distally and end with one large terminal spine. The male terminalia in ventral view appear as in Fig. 87.

The female wings are 2-2.1 mm long. The frons is shiny dark brown as are the terminal abdominal tergites. The clypeus is gray pollinose. The scutum is blackish gray and shiny with thin gray pollinosity laterally and sparce, small golden hairs. The subcosta is bare. The cerci are broadly rounded distally, bear many setae and in lateral view appear

Figure 84. Cephalic apotome and head spots of a *S. taxodium* larva.

Figure 85. Gular notch of a *S. taxodium* larva.

Figure 86. Pupal exuvium and cocoon of *S. taxodium*.

Figure 87. Male terminalia of *S. taxodium*.

Figure 88. Female terminalia of *S. taxodium*.

as one half of an oval almost as wide as the anal lobe. The genital fork is as in Fig. 88.

Distribution. *Simulium taxodium* is reported from southwestern Georgia (Snoddy and Beshear, 1968).

Life history. Little has been published on the life history of this species. Mature larvae and pupae were found to occur during early March but were absent in July and August (Snoddy and Beshear, 1968).

Ecology. Snoddy and Beshear (1968) report that bald cypress, *Taxodium distichum* Rich., is common where this black fly breeds.

Habits. Nothing has been seen in the literature concerning the habits of this species.

Florida observations.

Stream Parameters

	Width	Depth	pH	Temperature		Velocity	
Mean:	11.65 m	58.2 cm	6.09	20.4°C	(68.7°F)	.54 m/sec	(1.77 ft/sec)
Min:	.15	2	3.75	7.2	(45)	.15	(.5)
Max:	100	1000	7.55	29.2	(84.5)	1.78	(5.85)

Simulium taxodium has been found in 23 counties from west to south Florida at 39 locations (Fig. 89). This species has multiple generations each year and larvae and pupae have been collected during each month of the year. *Simulium taxodium* has been collected from a range of streams including streams with low pH reactions (4.3-4.5) such as Hatchet Creek at Site 20 and Site 99, the Alapaha River a large temporary flow which is usually just a dry white sand river bed in November or December of each year. Most preferred are streams with a pH reaction close to neutral (6.4-7.2) such as the intermittent, small Otter Creek (Site 135) which has been discovered not flowing during June and July as well as

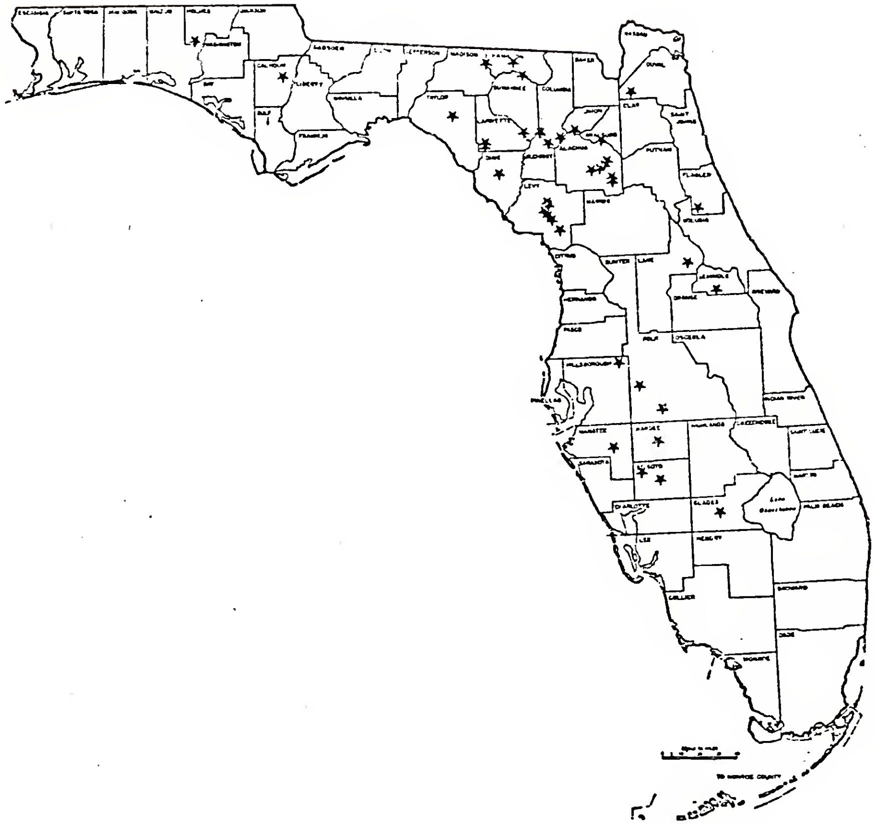

Figure 89. Collection locations for *S. taxodium* in Florida.

October and November and the permanent, larger, spring-fed Ichetucknee River (Site 59), both with abundant aquatic vegetation. Other flows with fairly neutral pH but little trailing vegetation, such as the Santa Fe River at Oleno State Park (Site 58) with moss-covered boulders a prominent feature in the flow and Blue Creek (Site 116) with considerable trapped twigs, leaves and other debris, consistently yielded *S. taxodium*. Cypress stands were notably present at the Ichetucknee River and Santa Fe River collection locations. Populations were found, generally, to be the largest when and where the flow was most swift, usually in the range .61-1.22 m/sec (2-4 ft/sec), which varied throughout the year in the various streams. At the Ichetucknee River (Fig. 90) *S. taxodium* was the predominant and almost exclusive black fly species present and was found year round on eel grass. At other locations *S. taxodium* immatures were collected from grass trailing from the banks, a wide variety of aquatic vegetation, vine, holly and water willow leaves, dead tree leaves, pine needles, twigs and rocks.

Simulium taxodium was associated with nine other black fly species in the streams and rivers (Table 3). Most frequently collected in association were *S. lakei* and *S. jenningsi*, two other members of the subgenus *Phosterodoros*. The widespread species *S. slossonae* and *S. tuberosum* were next most frequently collected with *S. taxodium*, followed by another *Phosterodoros* species, *S. jonesi*.

Florida specimens of *S. taxodium* differed from the descriptions in Snoddy and Beshear (1968) in a few ways. The larvae frequently had broad but pointed gular notches and the number of rays in each cephalic fan numbered between 50-60 rather than 38-42. The third petiole of the pupal respiratory organs was the longest of the four petioles but was

Figure 90. The Ichetucknee River, a *S. taxodium* collection site.

usually only 1/5 or 1/4 the length of the filaments themselves rather than the described length of 1/2 that of the filaments. The ventral plate was observed to expand distally in ventral view rather than remain fairly parallel-sided to the end and the plate in end view was not visible as a narrow sharp pointed structure but more rounded and tipped with a small knob.

One reared female of *S. taxodium* appeared to feed on a turkey in the laboratory as evidenced by a tiny feeding mark on the skin. When dissected three days later no blood was noticed in the gut of the fly and the ovaries lacked developed eggs.

Florida collection records for *S. taxodium*.

Alachua Co. 18) 1974: 12 Jan (L,P,A), 2 March (P), 11 April (L,P), 30 June (P), 24 July (L,P), 24 Aug (L,P), 22 Sept (L,P), 25 Oct (L,P), 23 Nov (L,P,A); 1975: 10 Jan (P), 31 Jan (L,P), 8 April (P,A), 11 May (P), 14 June (L), 1 Sept (P), 17 Oct (P), 10 Dec (L,P). 20) 1974: 12 Jan (L,P), 2 March (L,P), 12 April (L,P), 6 July (P), 29 Aug (P); 1975: 15 Jan (L), 31 Jan (L,P), 18 April (L,P), 26 July (P), 3 Oct (P). 21) 1974: 11 April (L,P), 14 Sept (L,P), 30 Oct (P); 1975: 24 Jan (L,P), 4 April (L,P), 27 Sept (L,P); 1976: 21 Jan (L,P), 29 Jan (L,P). 22) 1974: 28 Aug (L,P), 12 Sept (L), 7 Dec (L,P,A); 1975: 24 Jan (P), 4 April (L,P), 29 April (P), 5 Aug (P,A), 30 Aug (P), 17 Oct (P,A); 1976: 6 March (P). 24) 1974: 9 March (P), 25 May (L,P), 18 May (P). 39) 1975: 26 July (L,P).

Bradford Co. 45) 1974: 4 May (P), 16 Nov (P,A); 1975: 22 Nov (P).

Calhoun Co. 50) 1975: 11 June (P,A).

Columbia Co. 58) 1973: 25 Aug (L,P); 1974: 5 Jan (L,P,A), 23 Feb (L,P,C), 27 April (L,P,A), 1 July (L,P), 3 Aug (P), 21 Sept (P), 6 Nov (L,P); 1975: 12 Jan (L,P), 26 April (L,P,A), 21 June (L,P), 17 Aug (L,P), 22 Oct (P,C,A); 1976: 24 Jan (L,P). 59) 1973: 23 Sept (S,M,L,P,A), 6 Nov (S,M,L,P), 19 Dec (S,M,L,P); 1974: 23 Feb (S,M,L,P), 27 April (S,M,L,P), 7 May (S,M,L,P,A), 29 June (S,M,L,P), 3 Aug (S,M,L,P), 12 Aug (S,M,L,P,A), 21 Sept (S,M,L,P), 6 Nov (S,M, L,P); 1975: 12 Jan (S,M,L,P), 29 March (S,M,L,P), 21 June (S,M), 22 July (S,P,C), 27 Sept (S,M,L,P,C,A), 22 Nov (S,M,L,P). 62) 1975: 1 Nov (S,P).

Desoto Co. 65) 1974: 29 Nov (P,A); 1975: 22 March (P,A), 12 Sept (L), 22 Dec (P). 66) 1975: 12 Sept (P), 22 Dec (P).

Dixie Co. 69) 1975: 26 March (L).

Duval Co. 70) 1975: 12 March (L,P).

Flagler Co. 77) 1974: 7 Dec (P); 1975: 5 April (P).

Glades Co. 95) 1975: 17 June (P,C).

Hamilton Co. 99) 1974: 8 Aug (L); 1975: 26 April (L), 23 June (P), 3 Aug (L,P), 24 Oct (P,A).

Hardee Co. 104) 1975: 21 Dec (L).

Hillsborough Co. 113) 1975: 22 March (L,P), 27 May (L,P,A), 11 Sept (L,P), 21 Dec (L,P).

Holmes Co. 116) 1974: 17 March (M,L), 6 Aug (P), 13 Oct (L,P); 1975: 28 March (L,P), 11 June (P), 6 Sept (M), 30 Dec (P).

Lafayette Co. 129) 1974: 16 March (P,A); 1975: 26 March (L,P), 23 Aug (P), 28 Dec (P,A). 130) 1974: 12 Oct (L,P); 1975: 26 March (P), 10 June (P), 23 Aug (P), 28 Dec (P).

Lake Co. 170) 1975: 3 July (P).

Levy Co. <u>135</u>) 1973: 7 Oct (P); 1974: 2 March (L), 27 April (L,P,C), 3 Aug (L,P), 15 Sept (P); 1975: 31 Jan (P,A), 23 March (L,P,A), 17 May (P), 1 Sept (L,P), 30 Oct (P), 21 Dec (L,P). <u>136</u>) 1973: 30 Oct (P), 29 Dec (L); 1974: 2 March (L,P), 18 June (P), 15 Sept (P), 2 Nov (L,P); 1975: 31 Jan (P), 23 March (L), 1 Sept (P), 30 Oct (P). <u>137</u>) 1973: 29 Dec (L,P); 1974: 2 March (L,P), 27 April (L,P), 3 Aug (L,P), 15 Sept (L,P), 2 Nov (P); 1975: 31 Jan (P), 23 March (L,P), 1 Sept (L,P), 30 Oct (P). <u>138</u>) 1975: 31 Jan (P). <u>139</u>) 1975: 11 Sept (L), 30 Oct (L,P), 21 Dec (L,P). <u>141</u>) 1975: 17 May (L,P), 8 July (P), 1 Sept (P), 30 Oct (L,P).

Madison Co. <u>151</u>) 1973: 19 Dec (P); 1974: 10 Nov (L,P); 1975: 1 Feb (L), 25 April (L,P), 23 June (L,P,A), 3 Aug (P,A), 24 Oct (L); 1976: 14 Feb (P).

Manatee Co. <u>157</u>) 1975: 12 Sept (L,P), 22 Dec (L,P).

Polk Co. <u>181</u>) 1974: 29 Nov (L); 1975: 22 March (P), 21 Dec (P,A). <u>182</u>) 1975: 11 Sept (P), 21 Dec (L,P).

Seminole Co. <u>202</u>) 1974: 12 May (L); 1975: 15 March (P).

Suwanee Co. <u>207</u>) 1973: 23 Sept (P,A). <u>208</u>) 1974: 10 Nov (L); 1975: 1 Feb (L), 26 April (L,P); 1976: 24 Jan (P).

Taylor Co. <u>213</u>) 1974: 14 June (P), 5 Oct (P); 1975: 17 Jan (P), 26 March (L,P).

Union Co. <u>217</u>) 1974: 4 May (L,P), 6 July (L,P), 24 Aug (P), 9 Oct (P), 6 Nov (P); 1975: 22 July (P,A).

<u>*Simulium (Psilozia) vittatum* Zetterstedt</u>

Simulia vittata Zetterstedt, 1838, Insecta Lapponica, 1838-1840: 803 (female).

Simulium tribulatum Lugger, 1897, Minn. State Entomol. Rep. 2: 179 (female, male, larva, pupa).

Simulium glaucum Coquillett, 1902, Proc. U.S. Nat. Mus. 25: 97 (male).

Simulium venustoides Hart, 1912, in Forbes. Ill. State Entomol. Rep. 27: 42 (male only).

Psilozia groenlandica Enderlein, 1936, Gesell. naturf. Freunde, Sitzber.: 114 (female).

Simulium asakakae Smart, 1944, Roy. Entomol. Soc. London, Proc. (B) 13: 131.

Simulium vittatum — Stone, 1964, Conn. State Geol. and Natur. Hist. Surv. Bull. 97: 40 (female, male, larva, pupa).

Simulium vittatum — Stone and Snoddy, 1969, Auburn Univ. Agr. Exp. Sta. Bull. 390: 29 (female, male, larva, pupa).

Taxonomy. Zetterstedt (1838) first described a female of this species from Greenland. Stone (1964) indicates the holotype is in the University of Lund, Sweden. Peterson (1965) described the female specimen at the University of Lund and designated it as the lectotype in the event other of Zetterstedt's specimens materialize. Lugger (1897) described a male, larva and pupa for this species under the name *S. tribulatum*. Davies et al. (1962) suggest that this widely distributed black fly may be a complex of two species. Pasternak (1964) examined the larval salivary gland chromosomes of *S. vittatum* specimens from Alaska, Canada, and New England and concluded that no sibling species existed but that the species was very polymorphic due to exploitation of many niches.

Description. Wu (1930) describes *S. vittatum* eggs as .25 mm long, .15 mm broad and .14 mm high. The larvae are 8-9 mm long when fully

developed. The head capsule in preservation is light yellow with brown head spots. There is sometimes much infuscation about the head spots (Fig. 91). The gular notch is widest at its base, a little wider than long and is bluntly rounded or broadly pointed at its apex (Fig. 92). The subesophageal ganglion is dark colored, subtriangular in shape. The cephalic fans possess about 41-50 rays and have about 22 primary spines widely separated from each other along the ray with about 12 secondary spines in between each pair of primary spines. The submentum has especially prominent medial and lateral teeth and the margins of the submentum beyond the lateral teeth are conspicuously serrate. There are four to five long and additional short setae on each side of the submentum. The antenna is rather short with two white spots. In alcohol the abdomen is gray or greenish-gray to gray black with lighter intersegmental areas, while in nature the larva normally appears dark gray black. The anal gills appear as three, thick, simple lobes and anal tubercles are absent or very small.

The pupa is located in a slipper-shaped, well-woven cocoon with an anterior edge which slopes back ventral to dorsal and is 3-3.5 mm long. There are 16 filaments in each respiratory organ arranged in 8 pairs which arise at various distances from the base (Fig. 93).

The adult male is velvety black with a scutum that bears two anterior bright spots on the sides of a wide medial dark stripe which widens posteriorly and from the posterior sends two curved wide dark projections forward. The distimere is short, stout and bears 3-4 terminal teeth (Fig. 94). The ventral plate in end view is broadly triangular.

The wings of the female are 3-3.5 mm long. The female is ash gray

Figure 91. Head spots of a *S. vittatum* larva.

Figure 92. Gular notch of a *S. vittatum* larva.

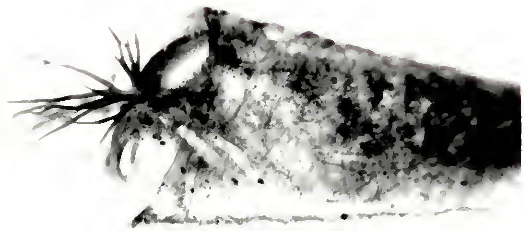

Figure 93. *S. vittatum* pupa and cocoon.

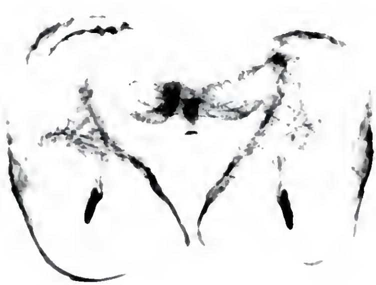

Figure 94. Terminalia of a *S. vittatum* male.

Figure 95. Scutum of a female of *S. vittatum*.

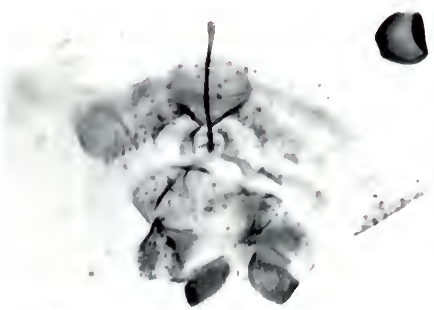

Figure 96. Terminalia of a female of *S. vittatum*.

in color with a silvery gray frons and gray clypeus. The scutum bears 5 conspicuous longitudinal brown marks, the outer pair on each side being almost spots and the middle mark essentially a solid line the length of the scutum (Fig. 95). There is a bold black and gray pattern on the abdomen. The genital fork appears as a widespread-Y with prominent ventral and dorsal projections distally on each arm (Fig. 96).

Distribution. In the U.S. *S. vittatum* has been reported from Alaska (Stone, 1952), Washington (Corredor, 1975), California (Hall, 1974), Utah and Idaho (Twinn, 1938), Kansas (Emery, 1914), Wisconsin (Anderson and Dicke, 1960), Minnesota (Nicholson and Mickel, 1950), New York (Stone and Jamnback, 1955), Delaware (Sutherland and Darsie, 1960a), New Jersey (Crans and McCuiston, 1970), Rhode Island (Dimond and Hart, 1953), Connecticut (Stone, 1964), Pennsylvania (Frost, 1949; Eckhart and Snetsinger, 1969), Massachussetts (Holbrook, 1967), the Tennessee River Valley (Snow et al., 1958), Maryland (Tarshis, 1968), Virginia (Townsend and Turner, 1976), South Carolina (Garris et al., 1975) and Alabama (Stone and Snoddy, 1969). The latter authors indicate *S. vittatum* occurs from Alaska and Greenland south to California, Texas, Louisiana, and Georgia and mention that it has not been located below the 29°N latitude with the southernmost records being Uvalde, Texas, and Baton Rouge, Louisiana. I record for the first time its presence in Florida.

Life history. Wu (1930) and Stone and Jamnback (1955) mention that *S. vittatum* females lay approximately 300 eggs in long gelatinous strings on leaves of aquatic plants, stones, and logs in streams. Davies and Peterson (1956) observed females of *S. vittatum* to fly facing the current of a sluiceway, drop down, touch their abdomens to the water's surface, release eggs, and then fly up and repeat the process. Peterson (1961)

found females oviposit by dipping their abdomens, in flight, to the water of a lake near a waterfall. Holbrook (1967) observed a *S. vittatum* female land on a solid object sticking out of the flow, turn, back into the water and lay eggs one inch below the surface. Stone and Snoddy (1969) indicate females will oviposit eggs on almost any substrate at or near the water surface. Wu (1930) found the eggs to hatch in nature at 20-22°C in 4 to 5 days and mentioned that hatching will also occur in standing water but that a longer time is required. Tarshis (1968) found *S. vittatum* eggs hatch in two days at 20-21°C in standing water. The eggs of *S. vittatum* were found to be unable to resist desiccation (Wu, 1930). The larval stage was found to last 13 to 17 days at 18-26°C (Wu, 1930). Anderson and Dicke (1960) report that *S. vittatum* larvae required three weeks for development in the summer in Wisconsin. Tarshis (1965) reported that pupation began 11 to 14 days after larvae were collected in the field. Snow et al. (1958) report the first broods of *S. vittatum* in the streams of Tennessee in late February and March. Larvae appear in March in Newfoundland (Lewis and Bennett, 1975). Garris et al. (1975) found larvae and pupae of *S. vittatum* only in May in Sumter Co., South Carolina. Wu (1930) found *S. vittatum* completed pupation in 3¼ to 5¼ days which agrees with the period of 3 to 4 days given by Anderson and Dicke (1960). Tarshis (1965) found adults began to emerge 1 to 2 days after pupation. Wu (1930) reported that a 50:50 ratio of males and females emerged from pupae she observed. Overwintering is reported for *S. vittatum* in the egg stage (Lewis and Bennett, 1973), in the larval stage (Dimond and Hart, 1953; Stone and Jamnback, 1955) and in the egg and larval stage (Anderson and Dicke, 1960). Two to three generations a year are reported in Newfoundland (Lewis and Bennett, 1973),

four generations in Rhode Island and New York (Diamond and Hart, 1953; Stone and Jamnback, 1955) and Stone and Snoddy (1969) report at least seven generations per year in Alabama.

Ecology. *Simulium vittatum* is reported to be one of the most abundant and widespread black flies in the irrigation canals of Saskatchewan and Alberta (Fredeen and Shemanchuk, 1960), in the streams and rivers of Wisconsin (Anderson and Dicke, 1960) and everywhere in Alabama except the coastal areas (Stone and Snoddy, 1969). Stone and Jamnback (1955) reported immature *S. vittatum* populations from dam faces and below lake outlets and large pools. Snow et al. (1958) found immatures on emergent water willow, and vines, grass and weeds trailing in the flow. Wolfe and Peterson (1959) found larvae initially on rocks but observed that they transferred to vegetation when the rocks became covered with algae. Sutherland and Darsie (1960a and b) recovered *S. vittatum* larvae from rivers and large streams in Delaware. Snow et al. (1958) found *S. vittatum* to be the most tolerant to silt of all the black fly species they observed and noted that adults would emerge successfully from silt-covered pupae on twigs and rocks. In Wisconsin *S. vittatum* larvae tolerated large amounts of eroded soil and other debris in the water courses (Anderson and Dicke, 1960). Immature populations of 2000 to 4000 larvae and pupae per .09 sq m (1 sq ft) were observed in Wisconsin (Anderson and Dicke, 1960). Fredeen and Shemanchuk (1960) report *S. vittatum* immatures from flows .03-1.5 m/sec (.1 to 5 ft/sec) with temperatures from 6.7°C (44°F) to 32°C (90°F) and pH from 7.3 to 9.6. Stone and Snoddy (1969) report the larvae will tolerate 0°C to 33°C water temperatures, flows from .15 to 1.83 m/sec (.5 to 6 ft/sec), and low pH and oxygen concentrations.

Jamnback (1973) reports transovarial transmission of microsporidian infections in 1 to 4% of surface sterilized eggs of *S. vittatum* observed in the lab. The fungus *Coelomycidium simulii*, often fatal to black fly larvae, was found in *S. vittatum* larvae in New York (Jamnback, 1973). Fredeen and Shemanchuk (1960) report three sites in Alberta where 95 to 100% of the larvae were parasitized by mermithid nematodes. Phelps and DeFoliart (1964) reported that three genera of mermithids which cause up to 50% larval mortality were found in the larvae of *S. vittatum* in Wisconsin and they record parasitism of adult *S. vittatum* by the genera *Gastromermis* and *Isomermis*. Molloy and Jamnback (1975) found that the mermithid *Neomesomermis* enters *S. vittatum* larvae by penetrating the integument and achieved, in lab trials, infections in 80% first instar and 64% second instar larvae and 41.9% mortality.

Habits. Mulla and Lacey (1976) found that younger *S. vittatum* larvae fed at a faster rate than older larvae. Bradbury and Bennett (1974a) found black, blue and red were the most attractive colors for *S. vittatum* adults. Peterson (1961) observed mating swarms of males 1.83-2.4 m (6-8 ft) above the lip of a waterfall. Hocking (1953) found that *S. vittatum* flies 258 cm/sec (5.8 mph) and exhibits continuous flight down to a temperature of 12.8°C (55°F) and intermittent flight to 8.9°C (48°F). Davies and Peterson (1956) and Stone and Snoddy (1969) record that males and females of *S. vittatum* are often collected from blossoms near a breeding site. Adults of *S. vittatum* were captured in light traps from May to September in Pennsylvania (Frost, 1949). In Alabama *S. vittatum* was captured in a modified New Jersey light trap with carbon dioxide as the attractant (Snoddy and Hays, 1966).

Wu (1930) found that development of the ova in *S. vittatum* is not

dependent on a blood meal but that the fat bodies provide enough nutrient. Lewis and Bennett (1973) report newly emerged females are autogenous for the first generation. Davies and Peterson (1956) reported that *S. vittatum* fed on deer in nature and on turkeys and ducks in the lab. Downe and Morrison (1957) found that *S. vittatum* fed on horses, cattle, and pigs. Snow et al. (1958) report attacks of *S. vittatum* severe enough to interrupt milking and plowing and drive work stock to cover and record cows being bitten on the udder while with horses and mules most black fly feeding occurred inside the ears. Fredeen (1973) reports that *S. vittatum* causes severe dermatitis in the ears of horses and cattle in Canada and Townsend and Turner (1976) report a similar situation with horses in Virginia. Davies et al. (1962) observed *S. vittatum* feeding on humans on two occasions. Jamnback (1969) indicates *S. vittatum* is a pest of livestock but rarely attacks man. In Pennsylvania *S. vittatum* is recorded as a non-biter but a pest of man (Eckhard and Snetsinger, 1969). Lewis and Bennett (1973) indicate *S. vittatum* is one of the more important pests of man in Newfoundland.

Florida observations.

Stream Parameters

	Width	Depth	pH	Temperature		Velocity	
Mean:	2.92 m	14.6 cm	6.1	19.2°C	(66.5°F)	.54 m/sec	(1.76 ft/sec)
Min:	.075	.5	4.35	10	(50)	.14	(.45)
Max:	15.25	90	7.4	27.8	(82)	1.77	(5.8)

In Florida *S. vittatum* was collected from 44 sites in 21 counties (Fig. 97). Included below is a record of specimens from the Withlacoochee River, Madison Co., which I examined but did not collect. *Simulium vittatum* is the seventh most widespread black fly in the state

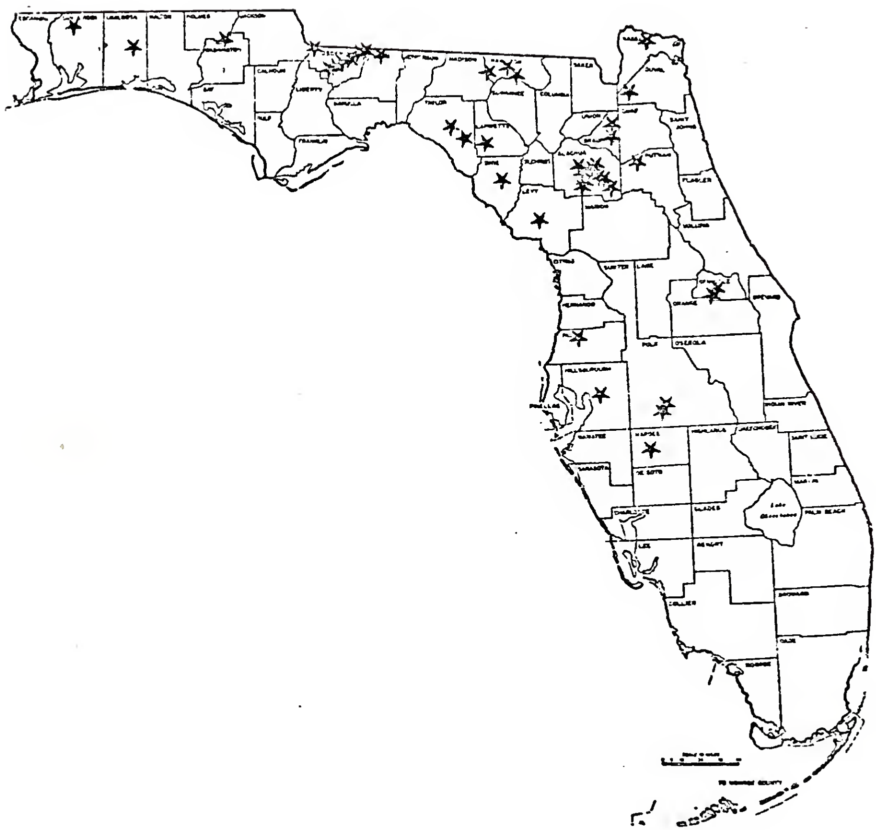

Figure 97. Collection locations for *S. vittatum* in Florida.

occurring in the western, central and more southern sections. There are five types of flowing water in which the immatures occur: 1) outflows of impounded water such as at spillways (Sites 15, 29, 33, 39,86, 132, 167, 177); 2) sand or mud-bottomed drainage ditches (130, 138, 196); 3) sandy or rocky streams with little vegetation (2, 6, 42, 47, 82, 90, 118); 4) streams clogged with vegetation (34, 72, 162); and 5) spring-fed rivers (181). The largest populations, approaching or exceeding 1000 larvae and pupae per .09 sq m (1 sq ft) were observed below ponds and lakes on spillways usually from January through April (Fig. 98). At Crestview (Site 167), the larvae formed a striking black carpet on the orange-colored sediment of the spillway. Larvae and occasionally even pupae appear in October in the streams and *S. vittatum* is present in most streams until middle or late June (Fig. 8). After June perhaps rising water temperatures or growth of unfavorable forms of algae cause *S. vittatum* streams to become unsuitable and populations disappear. In April 1975 a medium to heavy population of *S. vittatum* was present at Site 33 on the University of Florida in water with a temperature of 27.2°C (81°F). In July under similar flow conditions and a water temperature of 30°C (86°F) no *S. vittatum* immatures were found. Other factors accounting for the disappearance of *S. vittatum* could be early summer torrential rains which cause rushing stream conditions and increase the amount of abrasive particulate matter present possibly detrimental to *S. vittatum*; or, on the other hand, the long sunny, early summer days after the rains result in significant evaporation rates and, often, a decrease in stream flows at some sites, possibly too much to allow *S. vittatum* to remain present. There are no records of *S. vittatum* occurring in Florida in July, August, or September although aestivating

Figure 98. Site 167 at Crestview where *S. vittatum* was collected.

eggs are probably present. Many of the collection records listed below report only small, very young *S. vittatum* larvae which were found on only one or two of numerous visits to a site. Either the area where the real concentration of this species occurred was missed or, more probably, *S. vittatum* had difficulty surviving or building up large populations under those particular stream conditions. In sandy streams with sparse vegetation like Holman Branch (Site 82), at Quincy, the water was clouded with silt and only small numbers of *S. vittatum* along with a few *S. tuberosum* and *S. verecundum* were recovered. At another location, Site 18 — a flow to Newnan's Lake, *S. vittatum* was collected only once in nineteen visits over a period of two years, while six other species were collected there regularly. Perhaps this is an example of competitive exclusion. *S. vittatum* was found in the drainage ditches mentioned earlier but it was not collected in the main flows at the end of the ditches. The drainage flows were generally more shallow, less wide, warmer, and flowed slower than the main streams. Even in the drainage ditches and definitely in other flows, the largest number of *S. vittatum* immatures were found where the water channeled or was constricted and increased in velocity such a down a curved concrete ramp (Site 29), at the entrance to a pipe under the road (33), or where the stream poured off a concrete bridge support (111).

Larvae were collected from the following vegetational substrates in the streams: green filamentous algae, green and brown trailing bank grass, eel grass, dead tree leaves especially those of Magnolia, branches and twigs, soft aquatic plant stems, pine needles, bark, small aquatic bush leaves, roots, the leaves and stems of large elephant ear plants and trailing water willow leaves and branches. In addition larvae were

found on rocks, occasionally, and concrete, cardboard, black plastic, firm but gritty sand, and a wooden stake. Pupae were attached to many of the same substrates especially twigs, dead tree leaves, grass, green filamentous algae, and pebbles.

The summary of stream parameters above reveals that the immatures of *S. vittatum* were usually found in small to medium-sized streams with a moderate to swift flow that exhibited a slightly acidic to neutral pH reaction. The species association table (Table 3) reveals that *S. vittatum* was collected with ten other species in the streams of Florida. Most frequently *S. tuberosum* and *S. verecundum* were cohabitors with *S. vittatum*. Much less often *S. slossonae*, *S. lakei* and *S. jonesi* were found with *S. vittatum*. *S. decorum*, which often is found at the outflows of impounded water as is *S. vittatum*, had the advantage over *S. vittatum* at Shepard's Mill (Site 86) where *S. vittatum* was only collected once and *S. decorum* was found many times. In all other outflow situations *S. vittatum* populations greatly exceeded those of *S. decorum*. Only on one occasion during mid-March at the Crestview spillway (Site 167) were the numbers of *S. decorum* observed to approach those of *S. vittatum*.

In a few collections unusual forms of *S. vittatum* have been noted. In Hogtown Creek (Site 15) a white *S. vittatum* larva was collected but was judged to have just molted. Further downstream (Site 32) a group of *S. vittatum* larvae and some of *S. verecundum* were found all of which were whitish in color. The *S. vittatum* larvae appeared to have normal characteristics except for being unpigmented. Subsequent collections from the same site yielded slightly darker but still whitish gray *S. vittatum* larvae. Pupae have been found with 14 rather than 16 filaments in one of the respiratory organs and one pupa examined had respiratory organs

with 14 and 15 filaments. Only on two occasions were larvae observed with microsporidian infections. No mermithids have been noted in *S. vittatum* larvae.

Four adult female *S. vittatum* were captured during early May in a carbon dioxide-baited Manitoba trap near a stream containing *S. vittatum* immatures (Site 34). During March at the Tall Timbers Research Station in Leon County the ovipositing behavior of *S. vittatum* was observed. A silvery *S. vittatum* female, which was later captured and placed in alcohol, was observed at about 1700 hours to tap her abdomen while in flight to water pouring down a 45° concrete incline. The 1.2 m (4 ft) long concrete incline handled overflow from a large open swamp. The water on the spillway was 1-2 cm deep and flowed .61 m/sec (2 ft/sec). The female dipped down and touched the water two or three times as she flew up the incline. Other females flew very close to the water just above the lip of the spillway and may have been dropping eggs there also.

Florida collection records for *S. vittatum*.

Alachua Co. 1) 1974: 7 March (S). 2) 1974: 25 May (M). 7) 1974: 2 Feb (S), 7 March (S), 16 May (M,P); 1975: 12 Feb (S), 8 April (S). 11) 1973: 2 Nov (S). 13) 1974: 4 May (S). 15) 1973: 7 Dec (S,M,P); 1974: 2 Jan (S,M), 9 March (S,M,P,C), 13 April (S,M,P), 9 May (S,M,L,P,C), 7 June (S), 19 Oct (S,M), 16 Nov (S), 14 Dec (S,M,P); 1975: 31 Jan (S,M), 4 April (S,M,L,P,A), 13 May (S). 16) 1976: 13 March (S), 20 March (S,C). 18) 1975: 10 Jan (M). 22) 1974: 7 Dec (S). 29) 1974: 13 April (S), 9 May (S). 32) 1974: 2 Feb (S,P), 9 March (S,L), 13 April (P), 16 Nov (L); 1975: 10 Jan (S,M,P,A), 4 April (M). 33) 1974: 9 Jan (S,M,L,P), 14 Feb (S,M,C), 10 April (S,M,L,P), 18 May (S,M,L,P,A); 1975: 8 Jan

(S,M,L,P), 1 March (S,M,L,P,A), 30 April (S,M,L,P,A), 18 June (S,C), 22 Oct (P); 1976: 5 April (S,M,L,P,C,A). 34) 1974: 9 Jan (P,A), 13 March (S,M,P,C), 10 April (S,M,P), 3 May (A), 16 May (S,M,P); 1975: 8 Jan (S,M,P,C), 1 March (S,M,L,C), 30 April (S,M,L,P,C), 18 June (S,M,L,P); 1976: 5 April (S,M,L,P,A). 39) 1974: 2 Jan (S,M,L), 12 Jan (S), 14 Feb (S,M), 2 April (S,M,L,P,C), 16 May (S,M,L,P), 30 June (M), 25 Oct (S,M,L,P,A), 23 Nov (S,M,L,P,C,A); 1975: 10 Jan (S,M,L,), 1 March (S,M,L), 30 April (S,M,L,P,A), 19 Nov (S).

Bradford Co. 42) 1975: 5 April (S). 47) 1975: 26 April (S).

Dixie Co. 68) 1975: 17 Jan (S,L).

Duval Co. 72) 1974: 20 April (S,M); 1975: 12 March (S,M,L,P), 4 May (S,M).

Gadsden Co. 81) 1974: 17 March (S); 1975: 27 March (S). 82) 1974: 17 March (S,M,L,P); 1975: 18 Jan (S,L,P), 27 March (S), 10 June (S,M,P). 83) 1974: 17 March (S,M,L,P,C); 1975: 18 Jan (S,M,L,P,A), 27 March (S,M,L,P,C,A). 86) 1975: 18 Jan (P,A). 87) 1975: 27 March (S). 90) 1974: 17 March (S); 1975: 18 Jan (S,M,L), 27 March (S), 10 June (S), 29 Dec (S,M,L).

Hamilton Co. 100) 1976: 24 Jan (M). 101) 1975: 1 Feb (L).

Hardee Co. 102) 1975: 22 March (S,M,L,P).

Hillsborough Co. 111) 1975: 22 March (S,M,L,P,C,A).

Holmes Co. 118) 1975: 28 March (S,M,L,P,A), 11 June (S,M,L,C).

Lafayette Co. 130) 1974: 14 June (S,M,L,P,A); 1975: 26 March (S).

Leon Co. 132) 1974: 16 March (S,M,L,P,C,A).

Levy Co. 138) 1975: 31 Jan (M).

Madison Co. 152) 1967: 30 Nov (S,M,L — W. Beck).

Nassau Co. 162) 1974: 20 April (S); 1975: 12 March (P).

Okaloosa Co. 167) 1974: 18 March (S,M,L), 16 June (S,M,L,P), 13 Oct (S,M,L,P); 1975: 19 Jan (S,M,L,P,A), 28 March (S,M,L,P,A), 31 Dec (S); 1976: 18 April (S,M,L,P).

Pasco Co. 177) 1975: 23 March (S,C).

Polk Co. 181) 1975: 22 March (S). 183) 1975: 22 March (S).

Putnam Co. 186) 1975: 12 Feb (S).

Santa Rosa Co. 196) 1974: 18 March (S).

Seminole Co. 202) 1974: 21 March (L), 12 May (S,M,P), 28 Nov (S); 1975: 15 March (S,L,P), 27 April (S,M,L,P,C). 203) 1975: 15 March (S,P), 27 April (S,P).

Taylor Co. 210) 1973: 17 Dec (S,L). 213) 1975: 17 Jan (S).

Simulium (Simulium) decorum Walker

Simulium decorum Walker, 1848, List Dipt. Brit. Mus. 1: 112 (adult).

Simulium piscicidium Riley, 1870, Amer. Entomol. and Bot. 2: 367 (larva and pupa only).

Simulium venustoides Hart, 1912, in Forbes, Ill. State Entomol Rep. 27: 42 (female only).

Simulium decorum katmai Dyar and Shannon, 1927, Proc. U.S. Nat. Mus. 69(10): 31 (female).

Simulium ottawaense Twinn, 1936, Can. J. Res., D, 14: 146 (female, male, pupa).

Simulium decorum — Stone, 1964, Conn. State Geol. and Natur. Hist. Surv. Bull. 97: 44 (female, male, larva, pupa).

<u>Taxonomy</u>. Walker (1848) described an adult, apparently a female, from St. Martin's Falls, Albany River, Ontario. The holotype female is located in the British Museum, London, England (Davies et al., 1962). Stone (1964) mentioned that the relationship between *S. decorum* and supposedly synonymous European species like *S. noelleri* was still unclear. Stone and Snoddy (1969) suggest *S. argyreatum* Meigen, with which *S. noelleri* Frederichs is now considered synonymized, may be appropriate for the far north form in America which Dyar and Shannon called *S. decorum katmai*.

<u>Description</u>. The larva is 7.5-8 mm long, is gray in appearance in the preserved state and blackish gray in nature. The head capsule is a combination of light and dark brown with a dark brown fulvous area longitudinally on each side of a prominant median row of white head spots (Fig. 99). The cephalic fans each consist of about 57 rays. The gular notch is light colored, arrowhead-shaped, pointed distally, and extends about one half the distance to the teeth of the submentum (Fig. 100).

The pupa is located in a rather loose, coarse, cocoon 4-4.5 mm long and bears 8 thin filaments on each respiratory organ (Fig. 101).

The male wings are 2.5-3 mm long. The scutum is velvety black with many thin golden hairs and bears a faint silvery spot at each anterior lateral angle. The distimere is three times longer than wide and about twice as long as the basimere. The ventral plate in end view bears a median portion which is narrow and elongate (Fig. 102) and in lateral view displays a ventral keel.

The female wings are about 3 mm long. The base of the radius is bare. The frons and abdominal tergites are gray pollinose. The antennal

Figure 99. Head spots of a *S. decorum* larva.

Figure 100. Gular notch of a *S. decorum* larva.

Figure 101. *S. decorum* pupa and cocoon.

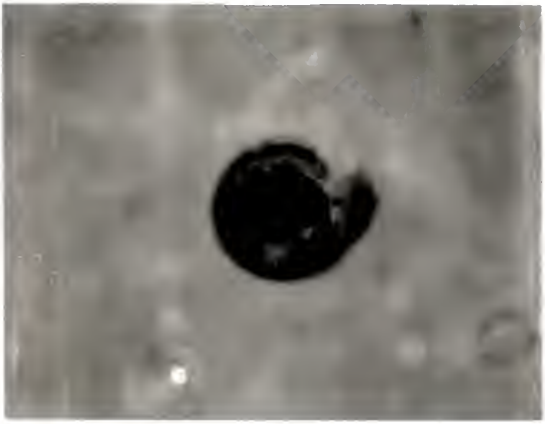

Figure 102. Male terminalia of *S. decorum*.

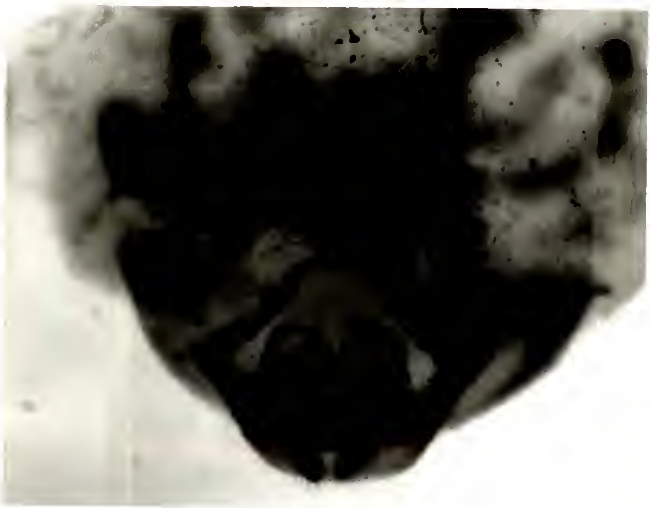

Figure 103. Female terminalia of *S. decorum*.

scape and pedicel are orange brown and the flagellum is dark brown. The fore tibia bear a dorsal elongate white patch. The legs are yellow brown with the segments darkening distally. The scutum is dark brownish gray in color and convex in lateral view. The genital fork is in the form of a broad Y with tapering arms that end in blunt, prominent, subapical projections (Fig. 103).

Distribution. Shewell (1957) lists *S. decorum* as Nearctic and widespread in northern America. Stone (1964) mentions this species is found from Alaska to Newfoundland and south to Oregon, Colorado, Arkansas, and Florida. Jones and Richey (1956) collected it in South Carolina, Snow et al. (1958) report it in their survey of the Tennessee River Valley and Stone and Snoddy (1969) found it commonly in central and northern Alabama.

Life history. In Ontario eggs overwinter and hatch during April (Davies et al., 1962). DeFoliart (1951) found the eggs hatch within seven days at 21.1°C (71°F). Tarshis (1968) found *S. decorum* eggs hatch in two days in standing water at room temperature (20-21°C). Stone and Snoddy (1969) report overwintering occurs as eggs, larvae or pupae. Anderson and Dicke (1960) mention first generation *S. decorum* larvae take 4 weeks to mature, second generation larvae take 3 weeks and pupae develop in 3 to 5 days. Davies et al. (1962) state that pupation occurs in mid-May and adult emergence takes place in late May. Abdelnur (1968) observed at least three cycles of pupation during 16-20 June, 22-25 July, and 11-19 August. Jones and Richey (1956) observed overlapping generations in South Carolina from February to May and suggest that with sufficient rain *S. decorum* breeds all summer with a generation every month. Stone and Snoddy (1969) indicate the initial mass emergence of adults

occurs during late April and a new generation is completed every five or six weeks thereafter until early November.

Ecology. Stone and Jamnback (1955) state that *S. decorum* larvae almost invariably occur on dams, at lake outlets or below large pools. Jones and Richey (1956) found 442 larvae per 6.45 cm^2 (1 in^2) on a dam spillway in April and Anderson and Dicke (1960) report populations of 1000-2000 larvae per .093 m^2 (1 ft^2) of substrate. Davies et al. (1962) found larvae in streams less than .3 m (1 ft) to over 4.6 m (15 ft) wide. Stone and Snoddy (1969) found *S. decorum* in streams with temperatures ranging from 0° to 33°C and currents up to 1.83 m/sec (6 ft/sec). Hudson and Hays (1975) found that in trough tests *S. decorum* larvae preferentially attached to substrates with particles in the .07 to .1 mm size range rather than substrates with smaller or larger particles. Larvae were found more abundantly in areas of dense plant parts such as the inner surfaces of root clusters. Ezenwa (1973) reported mermithid (*Neomesomermis*) and microsporidian (*Thelohania* and *Pleistophora*) infections in *S. decorum*. McKague and Wood (1974) found a granular SRIO Altosid formulation at .1 ppm concentration was 100% effective in halting adult emergence.

Habits. Females oviposit eggs in irregular masses while settled on vegetation, wood, concrete or rocks at the outlets of impounded water where a thin film of water flows over the substrate or, in more protected locations, may tap the surface of the water and drop one or more eggs into the water while in flight (Davies and Peterson, 1956; Davies et al., 1962). Adults may mate on the ground soon after emergence with the male approaching the female, crawling on her back, and curving his abdomen down to touch the tip of the female's abdomen (Davies and

Peterson, 1956). Davies and Peterson (1956) reported that *S. decorum* occasionally fed on humans, and in vials inverted over bare skin it fed on ducks, geese, crows and turkeys. Stone (1964) mentions this species is frequently a serious pest of horses, cattle and deer and suggest that it may attack birds. Jamnback (1969) lists *S. decorum* as mammalophilic but rarely a man-biter and mentions adults were found harboring developing trypanosomal stages. Stone and Snoddy (1969) found in Alabama that it attacks cattle and regularly bites man but was not considered a serious pest of humans.

Florida observations.

Stream Parameters

	Width	Depth	pH	Temperature		Velocity	
Mean:	7.37 m	7.7 cm	4.96	18.8°C	(65.8°F)	.82 m/sec	(2.7 ft/sec)
Min:	.1	.5	4.25	8.9	(48)	.3	(1)
Max:	36	60	7.0	26.7	(80)	1.83	(6)

Simulium decorum is recorded from 11 scattered locations in 5 Florida counties (Fig. 104). In almost every case the flow in which immatures of this species were found was immediately below or a short way downstream from a body of impounded water. A more specific search for black flies at locations such as on dams and at the outlets of lakes would most likely reveal a much wider or more uniform distribution for this species in Florida. At Shepard's Mill (Site 86), Chattachoochee (88), and Crestview (167) *S. decorum* was found on concrete spillways or dams where the flow reached 1.83 m/sec (6 ft/sec) at times. The width of the flows where *S. decorum* was found varied from 1 m or less at Site 33 on the Univ. of Florida campus and Site 132 at Tall Timbers to 12 and 36 m on the concrete at Shepard's Mill and the Chattahoochee Dam,

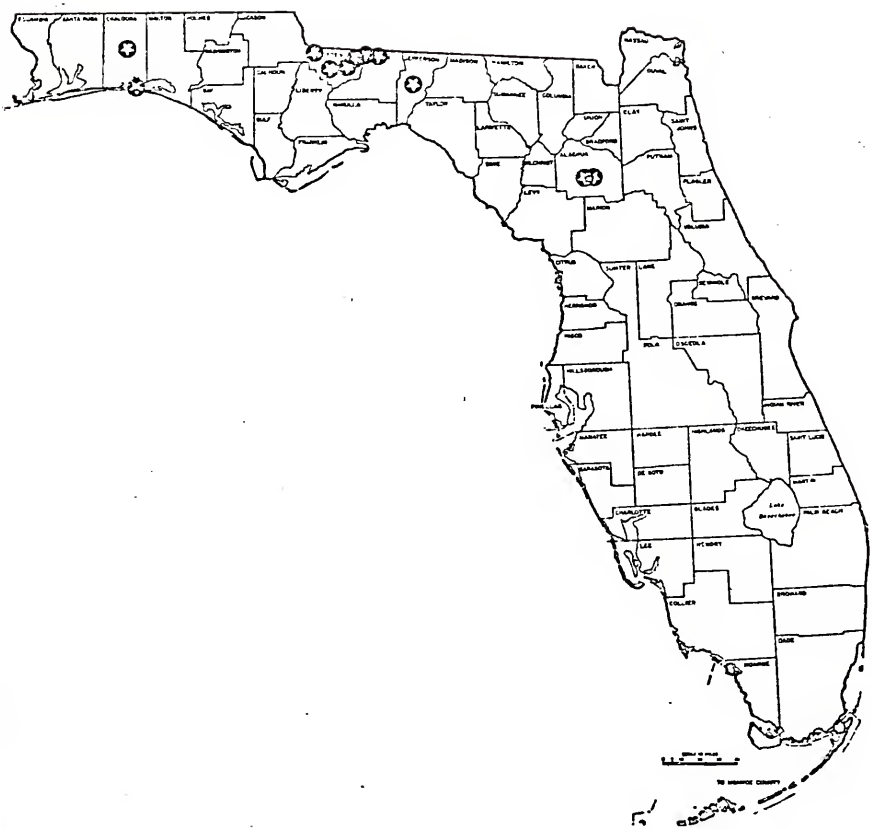

Figure 104. Collection locations for *S. decorum* in Florida.

respectively. The depth of the water at the collection sites was usually less than 10 cm. Larvae and pupae were found attached to pine needles, trapped grass, dead leaves, algae, concrete and orange-brown sediment. Collections at most locations were made from late December through mid June. At Shepard's Mill (Fig. 105) *S. decorum* was present in all stages all year. Water poured from Shepard's Pond over a series of 1.8 m wide and about 2 m high concrete and wood spillways next to each other onto a gently sloped, algae-covered, concrete run, passed under the road through three culverts and emptied into a large pool below. Heavy populations of larvae and pupae were found, especially during March, concentrated and lined up in narrow bands along irregularities in the concrete, more evenly distributed in large mats on the concrete and entangled in long thin green algae where the flow was a few centimeters deep and .3-1.5 m/sec (1-5 ft/sec) or faster. At this site during March females were observed flying back and forth at the crests of the spillways tapping their abdomens, presumably ovipositing, to the smooth water just before it plunged toward the concrete. Other females of *S. decorum* were removed during March and June from spider webs constructed on the supports of a few of the less active spillways.

Reflecting its presently known restricted distribution and preferred habitat *S. decorum* was associated with only five other black fly species in Florida's flows (Table 3). *Simulium vittatum* and *S. verecundum* were associated on nine and seven occasions, respectively. *Simulium tuberosum*, *S. jonesi*, and *S. notiale* were collected with *S. decorum* on six, three, and two occasions, respectively.

Simulium decorum females were identified as the flies involved in two human biting incidents, both in Okaloosa County: on 22 March in

Figure 105. Shepard's Mill, Site 86, where *S. decorum* was collected.

Crestview and 28 May in Destin. Female adults were captured on the wing in late March in Jefferson County and during early April in Alachua County.

Florida collection records for *S. decorum*.

Alachua Co. 33) 1976: 5 April (S). 34) 1976: 6 April (A).

Gadsden Co. 83) 1975: 27 March (M). 86) 1974: 17 March (S,M,L,P,A), 6 Aug (S,M,L,P,C), 13 Oct (S,M,L,P,C); 1975: 18 Jan (S,M,L,P,C,A), 27 March (S,M,L,P,C,A), 11 June (S,M,L,P,C,A), 24 Aug (S,M,L,P,C), 29 Dec (S,M,L,P,C,A). 88) 1975: 30 Dec (S,L); 1976: 17 April (S,M,L,P,A). 90) 1975: 18 Jan (S,M,L), 27 March (S,M,L,C). 91) 1975: 27 March (L).

Jefferson Co. 125) 1975: 26 March (A).

Leon Co. 132) 1974: 16 March (S,L,P,C).

Okaloosa Co. 167) 1974: 18 March (S,M,L), 16 June (S); 1975: 12 June (S,M,L); 1976: 22 March (A), 18 April (S). 224) 1976: 28 May (A).

Simulium (Simulium) tuberosum (Lundström)

Melusina tuberosa Lundström, 1911, Acta Soc. pro Fauna Flora Fenn. 34(12): 14 (male).

Simulium perissum Dyar and Shannon, 1927, Proc. U.S. Nat. Mus. 69(10): 43 (female, male).

Simulium vandalicum Dyar and Shannon, 1927, Proc. U.S. Nat. Mus. 69(10): 44 (male).

Simulium turmale Twinn, 1938, Can. Entomol. 70: 51 (male).

Simulium twinni Stains and Knowlton, 1940, Ann. Entomol. Soc. Amer. 33: 77 (male).

Simulium tuberosum, Stone and Jamnback, 1955, N.Y. State Mus. Bull. 349: 78 (female, male, pupa, larva).

Simulium tuberosum, Stone, 1964, Conn State. Geol. Natur. Hist. Surv. Bull. 97: 45 (female, male, larva, pupa).

Taxonomy. Lundström in 1911 in Finland first described a male of this species now known as *Simulium tuberosum*. Stone (1964) indicates that the syntypes are probably in the Kansallismuseo in Helsinki. Stone and Jamnback (1955) in their work on the black flies of New York State included descriptions of larva, pupa, and adults of *S. tuberosum*. Davies et al. (1962) and Jamnback (1969) suggest that *S. tuberosum* is a species complex of two or more undescribed forms. Landau (1962) separated four sibling forms of *S. tuberosum* based on variations in their salivary gland chromosomes. Stone and Snoddy (1969) mention two ecologically different forms in Alabama and suggest that if *S. tuberosum* in Europe is determined to be different from the form in North America, *S. perissum* Dyar and Shannon would be the most correct name for the southeastern United States species.

Description. Last instar larvae of *S. tuberosum* are 5.5-7.0 mm long. The abdomen is dark steel blue gray or reddish brown in color. The cephalic fans contain 37-42 rays. The head spots are darkish but indistinct and often appear as part of a fulvous pattern on the posterior portion of the cephalic apotome (Fig. 106). The gular notch is longer than wide, extends about one-half the distance to the submental teeth and is pointed or narrowly rounded at the apex (Fig. 107). The subesophageal ganglion is dark. The anal tubercles are small and inconspicuous.

Figure 106. The cephalic apotome of a *S. tuberosum* larva.

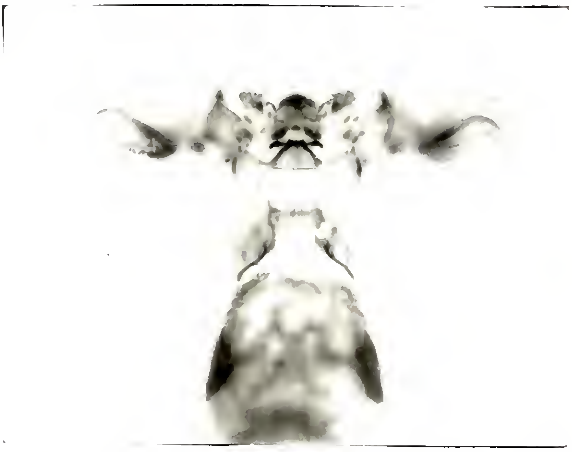

Figure 107. The gular notch of a *S. tuberosum* larva.

Figure 108. Pupa and cocoon of *S. tuberosum*.

Figure 109. Male terminalia of *S. tuberosum*.

Figure 110. Terminalia of a female of *S. tuberosum*.

The pupa is 2.5-3 mm long in a fairly uniformly textured, light-brown, slipper-shaped cocoon. The respiratory organ consists of six thin filaments about one-half the length of the pupa which rise in pairs from three short petioles (Fig. 108). The dorsal filaments are thicker than the ventral filaments. All pairs of filaments are fairly equally spaced from each other at the bases.

The males are velvety black in appearance with an oblique, silvery shiny patch at both antero-lateral angles of the scutum. Each curved, elongate distimere bears a number of spines on its basal lobe and the ventral plate in ventral view appears as in Fig. 109.

The female bears wings 2.5-3 mm long. The female is dark, gray and shiny in appearance with a shiny black frons and has hairs under the subcosta. Females possess a long dull white patch on each tibia which is not wider than one-third the width of the tibia. The ovipositor flaps or lobes are fairly parallel sided. The tarsal claws lack a basal tooth. The terminalia are as in Fig. 110.

Distribution. Shewell (1956) and Stone and Snoddy (1969) list *S. tuberosum* as circumboreal and holarctic, respectively. Davies et al. (1962) indicate that the type locality is probably Enontekis, Finnish Lapland, Finland. Ussova (1961) reported *S. tuberosum* from the U.S.S.R. while Grenier (1953) recorded it in France and Edwards (1915) and Smart (1944) noted its presence in England. Stone (1952) found *S. tuberosum* in Alaska. Stone (1964) lists the distribution of this species in the Nearctic region from Alaska and Greenland south to California, Texas and Florida.

Life history. Davies et al. (1962) indicate that in Canada the eggs overwinter and that there are three generations each year. Holbrook

(1967) found *S. tuberosum* to overwinter as eggs and to be multivoltine in Massachusetts. In Alabama overwintering occurs in the larval, pupal and adult stage (Stone and Snoddy, 1969). Davies et al. (1962) found that females emerge with undeveloped eggs and a moderate amount of stored nutrient. In Newfoundland the first generation larvae are present three to four weeks, the pupal stage lasts four to six days, the first generation adults emerge during mid-May and egg hatching of the second generation occurs two months later (Lewis and Bennett, 1973). Five or more generations are completed each year in Alabama (Stone and Snoddy, 1969).

Ecology. Sommerman et al. (1955) reported heavy (1500 larvae/ .09 sq m (1 sq ft)) populations of *S. tuberosum* in Alaskan streams and observed parasitism by mermithids in up to 65% of the individuals in some streams. Garris and Noblet (1975) report mermithid parasitism in South Carolina in *S. tuberosum* larvae from January through December ranged from 5 to 40%. Snow et al. (1958) found that *S. tuberosum* was the most frequently encountered black fly in the Tennessee Valley occurring in large numbers especially in mountain streams and in shallow, gravelly, well-exposed flows in lower areas. Ussova (1961) reported *S. tuberosum* from mountain rivers .5-2 m deep and found immatures attached to rocks and aquatic vegetation where the current was .5-1.2 m/sec in large and small creeks originating from lakes. Turbid, polluted water with an oxygen content less than 55% was rarely inhabited by *S. tuberosum*. Lewis and Bennett (1973) observed no substrate preference in *S. tuberosum* larvae and collected larvae from streams .5-20 cm deep with velocities from .27 to 1.34 m/sec and temperatures 6-18.5°C.

Habits. Davies and Peterson (1956) failed to observe *S. tuberosum*

females laying eggs on solid substrates and suggest that this species drops eggs freely into the water while flying. Wolfe and Peterson (1959) observed females of *S. tuberosum* land on the calm surface of stream water between stones and deposit about twenty eggs which sank to the bottom.

Downe and Morrison (1957) reported that *S. tuberosum* obtained blood from horses, cows, and chickens in a barn. Snow et al. (1958) collected *S. tuberosum* while it was feeding in the ears of horses and mules and crawling upon but not biting men about the head. Stone and Snoddy (1969) state that *S. tuberosum* is a serious pest of man and livestock in Alabama, feeding on the ears, udder, abdomen, and genitalia of cattle and readily biting people engaged in outdoor activities.

Florida observations.

Stream Parameters[1]

	Width	Depth	pH	Temperature		Velocity	
Mean:	2.24 m	22.93 cm	4.69	17.4°C	(66.3°F)	.52 m/sec	(1.7 ft/sec)
Min:	.03	1	3.5	7.2	(45)	.153	(.5)
Max:	10	100	7.6	29.4	(85)	1.53	(5)

Simulium tuberosum was collected from 97 locations in 30 counties in Florida (Fig. 111). This species is the second most abundant black fly in the state and is present in some streams all year round. The distribution of *S. tuberosum* includes locations in Escambia County in west Florida, points all across the coastal plains in the northern part

[1]The stream parameters above are compiled from figures gathered at over 50 sites involving more than 330 collections where at least three of the stages - S, M, L, or P - of *S. tuberosum* were found. Parameters from sites where *S. tuberosum* was less well represented, such as the big rivers, are not included.

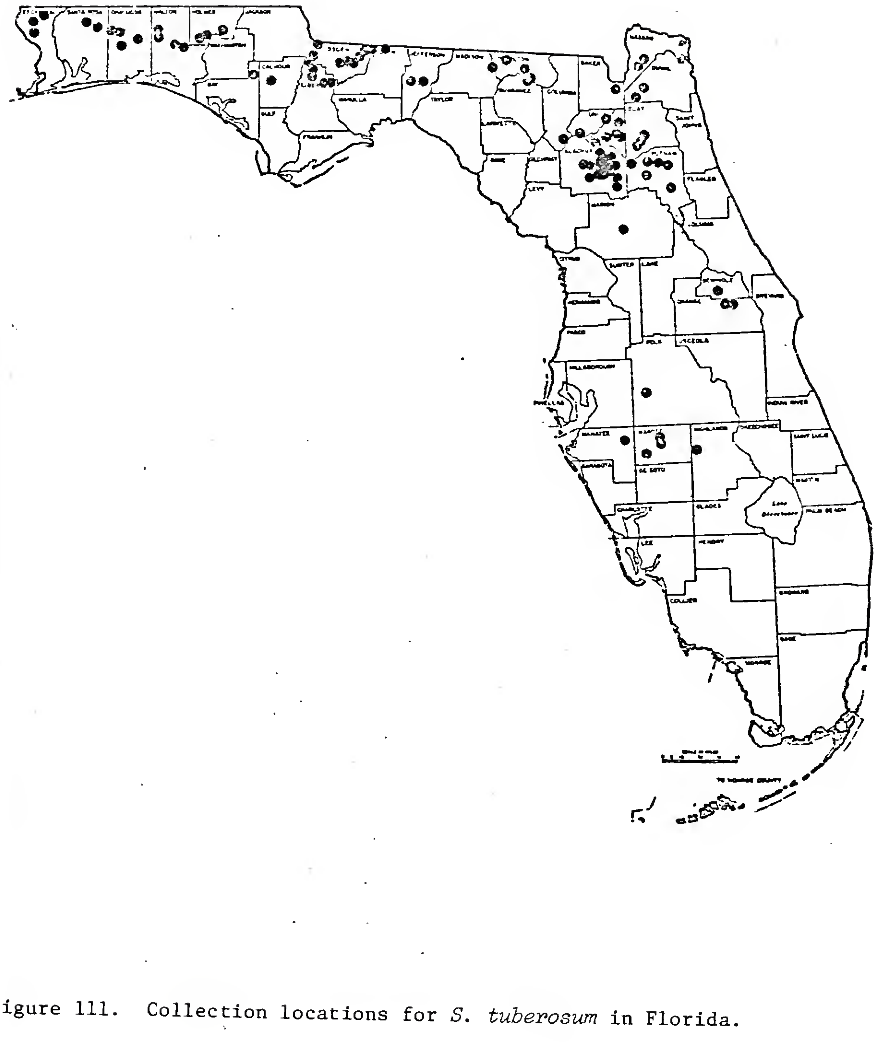

Figure 111. Collection locations for *S. tuberosum* in Florida.

of the state, generally high ground in the north central area and records as far south as Highlands County and Hardee County just northwest of Lake Okeechobee. This species was not collected in many counties bordering the Gulf of Mexico although a considerable number of collecting attempts were made for example in Levy County and Taylor County. In the counties of Alachua, Clay, Gadsden and Liberty *S. tuberosum* was found in many of the streams checked.

Simulium tuberosum was found occasionally in large rivers such as the Santa Fe (Site 58), the Blackwater (163), the Withlacoochee (151) and the Suwanee (208) however only in small numbers. Occasionally, in the spring, in the shallow flows of concrete drainage ditches near larger main streams such as at the Patuxent Research Station (Site 39) and at South Prong Black Creek (57) *S. tuberosum* was the only species present and dense populations were encountered.

There are two general types of streams, however, where *S. tuberosum* was usually present all year and reached its greatest numbers: small, sandy flows with little vegetation and small flows with considerable vegetation. The first category involves streams where *S. tuberosum* was usually the sole or at least the predominating species present and reached moderate to heavy numbers. These streams include Blues Creek (Site 2), a stream at the Devil's Millhopper (3), Gold Head Branch (55), Rock Creek at Torreya State Park (148) and Adam's Mill Creek (220). In these shallow, 1-3 m wide flows the substrate was sand or pebbles and sand and immatures were found attached to pine needles, dead leaves, roots, pebbles, rocks, clay and, occasionally, some trailing grass, sedges or other green aquatic vegetation. Most of these flows occurred in wooded, shaded situations.

The largest populations of *S. tuberosum* were discovered in streams of the second category with much vegetation such as the small flow at Site 8, Hatchet Creek (17), Site 56 (Fig. 112), West Minnow Branch (144), Thomas Creek (159) and Palmetto Branch (187). These streams were normally not more than 1 m wide or 30 cm deep. Abundant trailing grass, or large leafed and small leafed aquatic vegetation was located in the flow and it was on this flora plus trapped dead leaves that heavy numbers of *S. tuberosum* were found.

Another favored habitat of *S. tuberosum* was on the concrete of highway culverts where the stream poured off to continue its journey below. Good populations of immature *S. tuberosum* were found on the concrete, algae, leaves and other trapped debris in such situations at the tributary to Telogia Creek (Site 142) and Muddy Brook (Site 165).

As typical of most black fly species the immatures of *S. tuberosum* were usually found concentrated where the flow was the fastest. Site 148 at Torreya State Park illustrates this with the largest populations being found attached to the rock-hard surface of clay boulders over which a thin film of water poured down a meter high falls. At Site 8 during February 1974 large numbers of *S. tuberosum* were found concentrated in a 15 cm wide, 6 cm deep, grassy channel. At Palmetto Branch (Site 187) pools above and below the collection site restricted the current and black fly immatures to a sloped section a few meters long. In rushing Big Juniper Creek (Site 195) *S. tuberosum* was collected from algae-bearing rock-hard clay substrate, twigs and trailing water oak leaves where the water velocity often was 1.22 m/sec (4 ft/sec).

The stream populations of *S. tuberosum* were usually at their lowest during mid summer, began to increase by October, and reached maximum

Figure 112. Site 56 in Clay County, a collection spot for *S. tuberosum*.

numbers from January through May. This species is multivoltine in Florida and the seven or more generations each year overlap considerably.

Since *S. tuberosum* has adapted to a wide array of streams and is present all year long it was found in association with a great number of other black fly species. Fifteen other simuliid species were collected with *S. tuberosum* in Florida's streams and rivers (Table 3). Most frequently *S. tuberosum* was found with *S. jonesi* and *S. slossonae*, often on the same leaf, pine needle or other substrate. *Simulium lakei* and *S. congareenarum* were collected next most commonly with *S. tuberosum.*

Simulium tuberosum larvae were observed with retarded histoblast growth and bodies swollen with large white uniform masses or filled with many tiny white spheres which were interpreted as protozoan infections. Such infections were encountered in small larvae at six sites and, in larger populations, the infection rates ranged from 2.3% to 8.6%.

Mermithids were found in *S. tuberosum* larvae at ten locations and were observed in large or well-developed larvae once. Parasitism rates ranged from 1.3% (1 of 77 small larvae) to 39% (16 of 41) with most parasitism occurring in less than 10% of the small larvae collected. At one site (124) in Jefferson County mermithids were found parasitizing *S. tuberosum* larvae on 6 of 7 visits which covered all seasons of the year. It was at this site that 2 of 11 large larvae collected contained mermithid nematodes in the abdominal region. The respiratory histoblasts of these larvae were dark, well-formed, but about one-half normal size.

Two color variations in *S. tuberosum* larvae — a reddish-brown form and a steel-blue form — have been observed occurring alone in some streams and together at other sites. The gular notch appears more angular and pointed in the gray-blue form and more rounded apically in

the brown form. Variations in the ventral plate of the male have been noted for example whether the arms in ventral view are straight or curve outward and differences in the width of the main lower portion of the plate. From Site 102, Troublesome Creek, in Hardee County a male was reared which possessed a typical ventral plate but had claspers more broad than normal for *S. tuberosum*. *Simulium tuberosum* in Florida may actually be a species complex.

Adult females were captured in Manitoba traps (Table 4) and Malaise traps (Sites 2 and 25) during the spring. Reared females of *S. tuberosum* were given the opportunity to feed on turkeys in the laboratory on eight occasions from August to January. None of the females engorged on the birds.

Florida collection records for *S. tuberosum*.

Alachua Co. 1) 1973: 7 Dec (M), 13 Dec (S,P); 1974: 2 Feb (S,M,L,P), 7 March (S,M,L,P,A), 12 April (S,M,L,P), 6 July (S), 20 July (S); 1975: 10 Jan (S,M,L,P,C,A), 12 Feb (S,M,L,P,C,A), 4 April (S,M,L, P,C,A), 10 May (P,C), 15 July (L), 9 Nov (S); 1976: 21 Jan (S,M,L,P,A). 2) 1973: 25 Aug (S,M,L,P), 11 Oct (M,P,A); 1974: 2 Jan (S,M), 14 Feb (S,M,L,P), 2 April (S,M,L,P), 25 May (S,M,L,P), 30 June (S,M,L,P,C), 17 Aug (S,M,L,P), 21 Sept (S,M,P,C), 14 Dec (S); 1975: 10 Jan (S,M,L,P,A), 1 March (S,M,L,P,C), 19-20 and 21-23 March (A — G.B. Fairchild), 30 April (S,M,L,P,C), 26 May (P,C), 3 Oct (S,M,L,P,C,A), 10 Dec (S,M,L,P,C); 1976: 9-10 March (A — J. Glick). 3) 1973: 25 Aug (S,M,L); 1974: 2 April (S,M,P,A), 7 June (S,M, L,P), 14 Sept (S,M,L,P,C), 30 Oct (S,M,L,P,C); 1975: 10 Jan (S,M, L), 26 May (S,M,L,P), 15 July (S,M,L), 3 Oct (S,M,L,P,A). 6) 1973: 22 Sept (S,M,L,P,A), 16 Oct (S,M,L,P), 25 Oct (S,M,L,P); 1974:

2 Jan (S,M,L,P), 2 Feb (S,M,L,P), 7 March (S,M,L,P), 2 April (S,M,L,P), 18 May (S,M,L,P,C), 30 June (S,M,L,P,C), 30 July (S,M), 14 Sept (S,M,L,P,C), 10 Oct (S,M,P);1975: 15 Jan (S,M,L,P,C,A), 5 April (S,M,L,P,C,A), 18 May (S,M,L,P,C), 1 July (S,M,C), 16 Aug (S,M,L,C), 8 Oct (S,M,L,P,C), 6 Dec (S,M,L,P,C). 7) 1973: 13 Dec (S,M,L,P,C); 1974: 2 Feb (S,M,L,P), 7 March (S,M,L,P), 16 Nov (S,M,L,P,A), 14 Dec (S,P). 8) 1973: 5 Sept (S,M,L), 2 Nov (S,M, L,P); 1974: 14 Feb (S,M,L,P,C), 2 April (S,M,L,P), 13 May (S,M,C), 30 June (S,M,L,P), 3 Aug (S,M,L,P), 22 Sept (S,M,P), 25 Oct (S,M, L,P,A); 1975: 10 Jan (S,M,L,P), 12 Feb (S,M,L,P,C,A), 4 April (S,M,L,P,C,A), 18 May (P,C), 1 June (S,M,P,C,A), 18 June (S,M,L,P), 15 July (S,L), 25 July (P), 31 July (S,M,L,P,A), 30 Aug (S,C), 30 Sept (S,M,L,P,C), 9 Nov (S,M,L,P,C), 14 Dec (S,M,L,P,C,A); 1976: 6 March (S,M,L,P,C). 11) 1973: 2 Nov (S). 13) 1975: 8 Jan (S,M), 30 April (C). 15) 1973: 7 Dec (M,L); 1974: 2 Jan (S), 9 March (S,M), 16 Nov (S,M,L,P), 14 Dec (S,M,P). 16) 1976: 13 March (S,M,L,P), 20 March (S,M,L,P,C). 17) 1974: 2 Jan (S,M,L,P), 2 Feb (S,M,L,P,C), 7 March (S,M,L,P,A), 12 April (S,M,L,P,C), 5 May (S,M,L,P), 6 June (S,M,P,A), 1 July (S,M,L,P), 4 Dec (S); 1975: 11 Jan (S,P,C), 1 March (S,M,L,P,C,A), 30 April (S,M,L,P, C,A), 21 June (C), 16 Aug (S), 6 Dec (S,M,L,P). 18) 1974: 12 Jan (S,M,L,P,C,A), 2 March (S,M,L,P,C,A), 11 April (S,M,L,P,A), 25 May (S,M,L,P), 30 June (S,M,L,P,C), 24 July (M,L,P,C), 24 Aug (S,M,L,P), 22 Sept (M,L,P), 25 Oct (S,M,L,P), 23 Nov (S,M,L,P); 1975: 10 Jan (S,M,L,P,C,A), 31 Jan (S,M,L,P,C,A), 8 April (S,M,L,P,C,A), 11 May (S,M,L,P,A), 14 June (S,M,L,P,C), 17 July (S,M,L,P,C), 1 Sept (S,P,A), 17 Oct (S,P), 10 Dec (S,M,L,P,C,A). 20) 1974: 12 Jan

(P), 2 March (M,L), 12 April (P,C); 1975: 31 Jan (S), 18 April (S,M). 21) 1974: 2 March (S,M); 1975: 24 Jan (S,M), 4 April (S,M); 1976: 29 Jan (P). 22) 1974: 7 Dec (S); 1975: 24 Jan (S,L); 1976: 6 March (S). 24) 1976: 19 Feb (P). 25) 1975: 13 March and 15 March (A — J. Glick). 26) 1974: 13 April (S,M,L), 18 May (S,M,P,C). 27) 1974: 24 July (S,C). 28) 1975: 28 Oct (S), 9 Nov (S,M,L,P,C), 10 Dec (C); 1976: 30 Jan (S), 26 March (A). 32) 1974: 9 March (L,P), 16 Nov (S); 1975: 10 Jan (S,L,P, C,A); 1976: 21 Jan (S,M,L,P,A). 33) 1974: 9 Jan (P); 1975: 28 Nov (S). 34) 1974: 9 Jan (S); 1975: 28 Nov (S,M). 36) 1974: 12 April (S,M,L), 6 July (S,M); 1975: 15 Jan (S,M,L,P), 4 April (S,M,L,P,C,A), 22 May (S), 22 Oct (S,M,L,P,C,A). 39) 1974: 12 Jan (S,M), 14 Feb (P,C), 2 April (S), 25 Oct (S,M,P), 23 Nov (S,M,L,P); 1975: 10 Jan (S,M,L,P), 1 March (S,M), 30 April (S,M,L,P,C,A), 26 July (S,M,L,P), 27 Sept (S), 19 Nov (S,M,L,P,A); 1976: 21 Jan (S,M,L,P,C).

Bay Co. 40) 1975: 27 March (S,M,L,P), 11 June (S,M,L,P,C), 6 Sept (S,M,P,A), 30 Dec (S,P,C,A).

Baker Co. 41) 1974: 4 Jan (S,M,L,P), 1 June (S,M,L,P), 17 Sept (S,M, L,P), 6 Nov (S,M,L,P,C); 1975: 1 Feb (S,M,L,P,C,A), 26 April (S,M,P), 21 June (S,M,C), 17 Aug (S,M,L,P,C), 14 Nov (S,M,L,P).

Bradford Co. 42) 1973: 6 Oct (S,L,P); 1975: 19 Nov (S,M,L,P,A). 43) 1975: 5 April (P); 1976: 7 Jan (S), 21 Jan (S,M). 45) 1975: 26 Jan (S), 5 April (S). 47) 1974: 20 July (S), 21 Sept (S,M,P); 1975: 26 Jan (S,M,L,P,A), 26 April (S,M,L,P,A). 48) 1974: 3 Aug (P,A), 21 Sept (S,M); 1975: 5 April (S).

Calhoun Co. 49) 1975: 27 March (S,C,A), 6 Sept (S).

Clay Co. 55) 1973: 17 Nov (S,M,L,P); 1974: 4 Jan (S,M,L,P), 23 Feb (S,M,L,P), 4 May (S,M,L,P), 6 June (S,M,L,C), 20 July (S,M,L,P), 31 Aug (S,M,L,P,C), 25 Oct (S,M,L,P), 27 Nov (S,M,L,P,A); 1975: 16 Feb (S,M,L,P,C), 18 April (S,M,L,P), 21 June (S,L,P,C), 2 Aug (S,M,L,P,C), 19 Oct (S,M,L,P). 56) 1974: 4 Jan (S,M,L,P,C), 23 Feb (S,M,L), 4 May (S,M,L,P,C), 6 June (S,M,L,P,C), 20 July (S,M,L,P), 31 Aug (S,M), 25 Oct (S,M), 27 Nov (S,M,L,P,C); 1975: 16 Feb (S,M,L,P,C,A), 18 April (S,M,L,P,C), 21 June (S,M,P,C), 2 Aug (S,L,P,C), 19 Oct (S,M,L,P,C,A); 1976: 31 Jan (S,M,L,P,C,A). 57) 1974: 4 Jan (S), 23 Feb (S,L,P), 25 Oct (S), 27 Nov (S,M,P,A); 1975: 16 Feb (S,M,L,P,C), 18 April (S,M,L,P), 2 Aug (S), 19 Oct (L,P,A); 1976: 31 Jan (S,M,L,P,A).

Columbia Co. 58) 1975: 26 April (S).

Duval Co. 70) 1974: 4 Jan (L), 20 April (S,M,P,C); 1975: 12 March (P,C,A), 4 May (S,M,L,P,C,A), 1 July (S), 17 Aug (M,L,P,C,A); 1976: 14 Feb (S,M,L,P,C). 72) 1974: 5 Jan (S), 20 April (P); 1976: 14 Feb (M).

Escambia Co. 73) 1974: 18 March (S), 16 June (S,M,P), 7 Aug (S,L,P), 14 Oct (S,L,P,A); 1975: 19 Jan (S,P,C,A), 28 March (S,M,L), 12 June (S,M), 7 Sept (C), 31 Dec (S). 74) 1974: 16 June (S), 7 Aug (S), 14 Oct (S); 1975: 19 Jan (S,L), 28 March (S,M,P,A), 12 June (S), 31 Dec (S); 1976: 18 April (S,M,P). 75) 1974: 14 Oct (M); 1975: 28 March (S,P,C,A), 12 June (S,L,P), 31 Dec (S,P,C).

Gadsden Co. 81) 1973: 18 Dec (S,M,L); 1974: 17 March (S,M,L,P,C), 15 June (S,M,L,P,A); 1975: 27 March (S,M,L,P,C), 10 June (C), 5 Sept (S). 82) 1974: 6 Aug (M); 1975: 18 Jan (S), 27 March (S,M,L), 10 June (S,M,L), 23 Aug (S,M), 29 Dec (S,M). 83) 1973:

18 Dec (S); 1974: 17 March (S,M,L,P,C); 1975: 18 Jan (S,M,L,P,A), 27 March (S,M,L,P,A), 10 June (S,M,L,P), 24 Aug (S), 29 Dec (M). 85) 1975: 30 Dec (S). 87) 1974: 15 June (M), 5 Aug (S,L), 12 Oct (S,M,L,P); 1975: 18 Jan (S,M,L,P,A), 27 March (S,M,L,P,C), 10 June (S,M,P,C), 5 Sept (S,M,L), 29 Dec (S,M,L,P,C,A). 88) 1975: 30 Dec (M,L,C); 1976: 17 April (M,L,P,C,A). 89) 1975: 30 Dec (S,M,L,P,C,A). 90) 1974: 17 March (S,M,L), 15 June (S,M,L,P), 6 Aug (S,M,P), 13 Oct (S,M,L); 1975: 18 Jan (S,M), 27 March (S,M, L,P), 10 June (S,M,L,P), 23 Aug (S,L), 29 Dec (S,M,L,P,C,A). 91) 1975: 18 Jan (S,M), 27 March (S,M,L,P,C,A), 29 Dec (S).

Hamilton Co. 99) 1974: 8 Aug (M); 1975: 1 Feb (S), 26 April (S,M); 1976: 14 Feb (S). 100) 1974: 8 Aug (S,M,L,P,C), 17 Sept (S,M,L, P,C,A), 10 Nov (S,M,L,P); 1975: 23 June (S,M,L,P,C), 24 Oct (S,M, L,P,C,A); 1976: 24 Jan (S,M,L,P). 101) 1975: 1 Feb (S,M,L,P,A), 26 April (S,P).

Hardee Co. 102) 1975: 22 March (S,M,L,P,C), 27 May (M,P,C,A), 22 Dec (S,M,L,P,A). 103) 1975: 22 March (S,M,L,P,C,A), 11 Sept (S,M,P,A), 22 Dec (S,M,L,P,C). 104) 1975: 11 Sept (S,M,L,P,C), 21 Dec (S,M, L,P,C,A).

Highlands Co. 109) 1974: 24 March (S,M,L,P), 12 May (S,M,L,P,C,A), 23 June (P,A), 29 Nov (S,L,C); 1975: 21 March (S,M,L,P,A), 27 May (S,M,L,P,C), 9 Aug (S,M,L,P,C), 15 Oct (S,L).

Holmes Co. 116) 1975: 19 Jan (S,M,L,P,A), 28 March (S,M,P,C), 6 Sept (P). 117) 1975: 28 March (S,P,C), 11 June (S,M,L), 6 Sept (C), 30 Dec (M). 119) 1975: 19 Jan (S), 28 March (S), 11 June (S,M,L, P,A), 30 Dec (P,C,A); 1976: 18 April (S,M,L,P,C,A).

Jefferson Co. 124) 1974: 16 March (S,M,L,P), 12 Oct (S,M,L,P); 1975:

17 Jan (S,M,L,P,A), 26 March (S,M,L,P,C), 10 June (S,M,L,P), 23 Aug (S,M,L,P,C), 29 Dec (S). 125) 1974: 5 Aug (S,M,L,P), 12 Oct (S,M, L,P,A); 1975: 17 Jan (S,M,L,P), 26 March (S,M,L,P,C,A), 10 June (S,M), 23 Aug (S,M,C), 29 Dec (S,M).

Leon Co. 131) 1974: 16 March (S,M,L,P,C); 1975: 26 March (S,M,L,P), 10 June (P,C), 23 Aug (S,M,L,P,C), 29 Dec (S,M,P,C).

Liberty Co. 142) 1974: 17 March (S,M,L,P,C,A), 15 June (S,M,), 13 Oct (S,M,L,P,A); 1975: 18 Jan (S,M,L,P,A), 27 March (S,M,L,P,C,A), 11 June (S,M,L,P,C), 24 Aug (S,M,L,P,C), 30 Dec (S,M,P,C). 144) 1973: 18 Dec (S,M,L,P); 1974: 17 March (S,M,L,P,C,A), 15 June (S,M,L,P), 6 Aug (S,M,P,C), 13 Oct (S,M,P,A); 1975: 18 Jan (S,M, L,P,C), 27 March (S,M,L,P,C,A), 11 June (S,P,C), 24 Aug (S,M,L,P, C,A), 30 Dec (S,P,C,A). 146) 1975: 18 Jan (M), 30 Dec (S,M). 147) 1974: 17 March (S,M,L,P,A); 1975: 18 Jan (S,M,L), 27 March (S,M,L,P,C), 11 June (S,M,L,P,C), 24 Aug (S,M,L,P,C), 30 Dec (S,M, L,C). 148) 1974: 15 June (S,M,L,P), 6 Aug (S,M,L,C), 13 Oct (S,M,L,P,A); 1975: 18 Jan (S,M,L), 27 March (S,M,L,P,C,A), 11 June (S,M,L,P), 6 Sept (S,M,L,P,A), 30 Dec (S,M,L,P,A).

Madison Co. 151) 1975: 1 Feb (P); 1976: 14 Feb (L).

Manatee Co. 155) 1975: 22 March (S,M,P,A), 27 May (S), 12 Sept (C), 22 Dec (S,P).

Marion Co. 158) 1952: 12 March (S,M — W. Beck).

Nassau Co. 159) 1974: 4 Jan (S,M,L,P,C), 20 April (S,M,L,P,C), 10 July (S,M), 24 Aug (S,M,L,P), 9 Oct (S,M,P,C), 4 Dec (S,M,L,P,C,A); 1975: 12 March (S,M,L,P,C), 4 May (S,M,L,P,C), 1 July (S,P), 26 Sept (S,M); 1976: 14 Feb (S,M,L,P,A). 160) 1974: 20 April (S,M, L,P,C); 1975: 12 March (S,M), 4 May (S,M,P,A), 26 Sept (P).

Okaloosa Co. <u>163</u>) 1974: 7 Aug (S); 1975: 19 Jan (S), 28 March (S), 31 Dec (P). <u>165</u>) 1974: 18 March (S,M,L,P,C,A), 16 June (S,M,L,P), 7 Aug (S,M,L,P), 14 Oct (S,M,L,P); 1975: 19 Jan (S), 28 March (S,M,L,P,C), 12 June (S,M,L,P,C), 31 Dec (S,L,P,C). <u>166</u>) 1974: 18 March (L,C), 14 Oct (S,M); 1975: 19 Jan (S,M,P,C), 28 March (S,M,C), 12 June (S). <u>168</u>) 1973: 13 March (S,P,A — K. Tennessen); 1975: 29 March (S,M,L), 12 June (S,M,L,P,C), 6 Sept (C).

Orange Co. <u>169</u>) 1974: 13 July (L,P,C), 28 Nov (S,M,L,P,C); 1975: 15 March (P,C), 31 Oct (S). <u>174</u>) 1947: 6 and 7 Feb (A — H.K. Gouck).

Polk Co. <u>182</u>) 1975: 22 March (S), 27 May (S,M).

Putnam Co. <u>185</u>) 1974: 14 Feb (P), 26 March (P), 23 Nov (P); 1975: 15 Jan (M,L). <u>186</u>) 1974: 19 Jan (L), 25 May (P); 1975: 12 Feb (S,M,P), 18 April (S,P,C). <u>187</u>) 1974: 19 Jan (S,M,L,P,C), 14 April (S,M,L,P,C,A), 25 May (S,M,L,P,C), 6 July (S,M,L,P), 17 Aug (S,M,L,P,C,A), 6 Oct (S,M,L,P,C,A), 23 Nov (S,M,L,P,C,A); 1975: 12 Feb (S,M,L,P,C,A), 18 April (S,M,L,P,C), 26 June (S,M,L,P,C,A), 25 Sept (S,M,L,P,A). <u>189</u>) 1975: 12 Feb (C), 26 June (P), 31 Oct (S). <u>191</u>) 1974: 19 Jan (M), 14 April (M); 1975: 5 April (S). <u>192</u>) 1974: 25 May (S,M,L,P,C), 6 Oct (S,M,L,P), 7 Dec (S,M,L,P,C,A); 1975: 5 April (S,M,L,P,C), 7 Aug (S,M,L,P,C,A), 31 Oct (S,M,L,P,C,A).

Santa Rosa Co. <u>195</u>) 1974: 18 March (S,M,L,P,C), 16 June (S,M,L,P,C), 7 Aug (S,M,L,P), 14 Oct (S,M,L,P,A); 1975: 19 Jan (S,M,L,P,A), 28 March (S,M,L,P,A), 12 June (S,M,L,P,A), 7 Sept (S,M,L,P,C), 31 Dec (S,M,L,P,C); 1976: 18 April (S,M,L,P,C). <u>196</u>) 1974: 18 March (S,M,L), 16 June (S,M,P), 7 Aug (S,M,L), 14 Oct (S,M,L,P,A); 1975: 19 Jan (S,M,L,P,C,A), 28 March (S,M,L,P,C), 12 June (S,M,L,P,C), 7 Sept (C), 31 Dec (S,P,C).

Seminole Co. 203) 1974: 21 March (S,M,L,P), 12 May (P), 28 Nov (P); 1975: 15 March (L,P,C), 4 July (S,P).

Suwanee Co. 208) 1976: 24 Jan (S).

Union Co. 216) 1975: 4 May (A). 217) 1974: 5 Jan (S,M,L,P), 23 Feb (S,M,L,P), 4 May (S,M,L,P,A), 6 July (S,L,P,C), 24 Aug (S,L,C), 9 Oct (C), 6 Nov (S,M); 1975: 12 Jan (S,L,P,A), 8 April (S,M,L, P,C), 1 June (S,C), 5 Oct (S,M,P,C).

Walton Co. 220) 1974: 18 March (S,M), 16 June (S,M,P), 7 Aug (S,M,L), 13 Oct (S,M,L,P); 1975: 19 Jan (S,C), 28 March (S,M,L,P,C), 11 June (S,M,L,P,C), 6 Sept (S,L,P,C,A), 31 Dec (S,M,P,C,A). 221) 1975: 28 March (M). 222) 1974: 6 Aug (P,A). 223) 1975: 19 Jan (S,M, L,P,A), 28 March (S,M,L,P,C,A), 11 June (P,C,A), 6 Sept (S,P,C,A).

Simulium (Simulium) verecundum Stone and Jamnback

Simulium verecundum Stone and Jamnback, 1955, N.Y. State Mus. Bull. 349: 83 (male, female, larva).

Simulium verecundum, Davies, Peterson, and Wood, 1962, Proc. Entomol. Soc. Ontario 92: 125 (female, male).

Simulium verecundum, Wood, Peterson, Davies, and Gyorkos, 1963, Proc. Entomol. Soc. Ontario 93: 114 (larva).

Simulium verecundum, Stone, 1964, Conn. State Geol. and Natur. Hist. Surv. Bull. 97: 47 (male, female, larva, pupa).

Taxonomy. Stone and Jamnback (1955) first separated this species from the closely related *S. venustum* based primarily on characteristics of the male genitalia. The holotype male was collected in Monroe Co. Pennsylvania and is located in the U.S. National Museum. Stone (1964)

indicates *S. verecundum* may be the same as *S. argyreatum* Meigen as defined by Rubtsov but the true identity of *S. argyreatum* is under question and therefore *S. verecundum* should be retained as the name for the American specimens.

<u>Description</u>. The larva is 6-7 mm long and appears white when recently collected. The head spots are white against a light brown frontoclypeus (Fig. 113). The gular notch is elongate and rounded anteriorly (Fig. 114). The ventral tubercles are visible but not large.

The pupa is about 3.5 mm long. The respiratory filaments are 6 in number, almost 3 mm long, and are arranged in pairs. They are spread wide with the bases of filament pairs 3-4 and 5-6 diverging greatly from each other (Fig. 115). The cocoon is brown, slipper-shaped and well-woven.

The male is velvety black in appearance with two silvery, lateral patches on the anterior third of the scutum which connect along the sides of the scutum to a shiny silvery band at the posterior. Each tibia bears a bright white patch. The distimeres are elongate, wide and fairly flat. The arms of the ventral plate in ventral view curve inward slightly toward each other and the median portion or apex of the plate is narrow and elongate in end view (Fig. 116).

The wings of the female are 3 mm long. The frons is shiny black. There are ten or more hairs on the underside of the subcosta. The tarsal claw lacks a basal tooth. Each fore tibia has a wide, bright white patch which extends more than three-fourths of the length of the tibia. The ovipositor lobes are dark, sclerotized and distinctly concave on their inner margins, creating an oval space between them (Fig. 117).

<u>Distribution</u>. Stone and Snoddy (1969) mention *S. verecundum* occuring

Figure 113. The head spots of a *S. verecundum* larva.

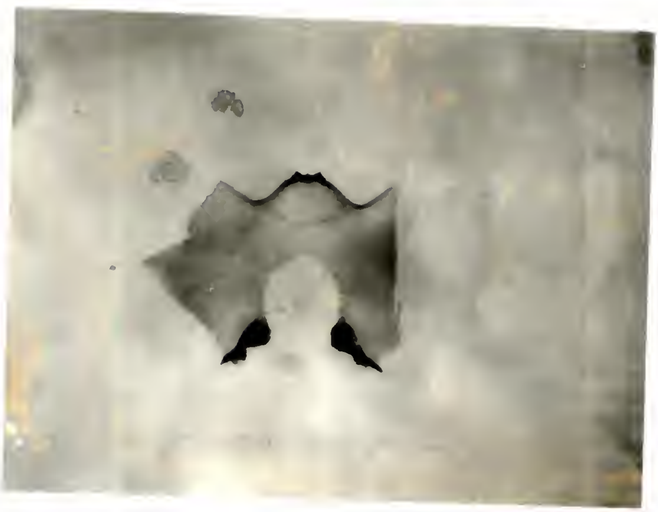

Figure 114. The gular notch of a *S. verecundum* larva.

Figure 115. The pupa and cocoon of *S. verecundum*.

Figure 116. Male terminalia of *S. verecundum*.

Figure 117. Terminalia of a female of *S. verecundum*.

from Alaska to Nova Scotia south to Washington, Wyoming, South Carolina and Alabama and they add that it is also found in Europe and Northern Asia.

Life history. Golini and Davies (1975) report that the eggs of *S. verecundum* are .228 mm long and .139 mm wide. Davies et al. (1962) in Canada and Holbrook (1967) in Massachusetts indicate that *S. verecundum* eggs overwinter. Larvae appear in late April or early May and adults are present by late May. Anderson and Dicke (1960) found immature stages of *S. verecundum* in Wisconsin from July to September. Lewis and Bennett (1973) report larvae develop in 3 weeks and pupae in 4-7 days. Snow et al. (1958) reared one male from a pupa collected in Mississippi on 30 June. Stone and Jamnback (1955) suggest that this species has two or three generations each year and Stone and Snoddy (1969) indicate it is multivoltine in Alabama. Lewis and Bennett (1974) report *S. verecundum* first appears in Newfoundland in early June and is present until late October completing three or four generations each summer.

Ecology. Davies et al. (1962) indicate that *S. verecundum* is generally found in larger streams over 3 m (10 ft) wide. Holbrook (1967) reports that 10 of 14 collections of *S. verecundum* were made below pond outlets and swamps. In Massachusetts, streams inhabited by *S. verecundum* were very small to about 2.5 meters wide. Snow et al. (1958) collected *S. verecundum* from a sandy-bottomed stream. Stone and Snoddy (1969) found this species to be uncommon in Alabama but collected it on the spillways of dams. Lewis and Bennett (1973) found immatures on all available substrates but they were most abundant on trailing vegetation late in the summer. Garris and Noblet (1975) report 23-27%

parasitism by mermithids in *S. verecundum* in South Carolina. Dove and McKague (1975) report .001-.1 ppm of Altosid reduced adult emergence of *S. verecundum* by 75-100% at temperatures between 10 and 25°C.

Habits. Golini and Davies (1975) report that *S. verecundum* females oviposit by first landing at the water's edge on trailing vegetation and then depositing an average of 417 eggs per female in large irregular masses sometimes five layers deep on vegetation. The females of *S. verecundum* were found to oviposit more frequently on green and yellow test strips in the flow than on strips of other colors.

Stone and Jamnback (1955) and Jamnback (1969) report *S. verecundum* does not commonly annoy or attack man. Lewis and Bennett (1973) indicate this species is anautogenous. Abdelnur (1968) captured females feeding on cattle.

Florida observations.

Stream Parameters

	Width	Depth	pH	Temperature		Velocity	
Mean:	2.76 m	25.8 cm	5.09	17.4°C	(63.3°F)	.45 m/sec	(1.49 ft/sec)
Min:	.15	1.27	3.75	6.7	(44)	.076	(.25)
Max:	36	166	7.0	27.8	(82)	1.53	(5)

Simulium verecundum is reported for the first time from Florida. *Simulium verecundum* was collected from 48 sites in 22 counties (Fig. 118). It was widespread across the northern half of Florida but was not found in collections south of Seminole County. *Simulium verecundum* was found in streams from mid-October through mid-June (Fig. 8). At Pine Barrens Creek (Site 74) on 14 October larvae and pupae were collected; at all other locations pupae were not found until after October. During June young and mature larvae and pupae were located. There are presently no

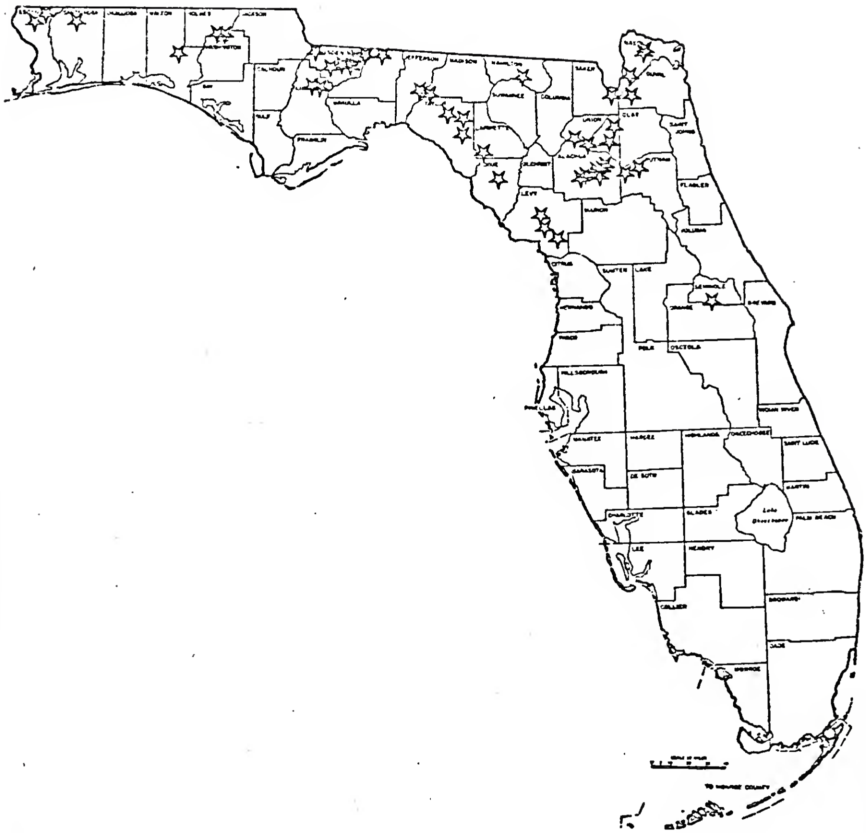

Figure 118. Collection locations for *S. verecundum* in Florida.

records of *S. verecundum* from Florida during July, August, and September. The large whitish and sometimes slightly yellow larvae of this species and the fairly large pupae were most abundant during January through April. Generally, small streams not more than a few meters wide were preferred by *S. verecundum*, such as Panther Creek, Site 223 (Fig. 119), although a good number of larvae and pupae were found at Pine Barrens Creek (Site 74) which in unflooded condition is about 10 meters across. Other favorable locations including Hatchet Creek (Site 17), Hogtown Creek (32), Hurricane Creek (83), and a tributary to the Aucilla River (153) illustrate that the suitable depth of the streams varied from about 20 cm to a meter. Optimum stream velocity ranged from .3 m/sec (1 ft/sec) to .76 m/sec (2.5 ft/sec). On one occasion a live pupa from which a female was reared was found in a trickle moving about .076 m/sec (.25 ft/sec). The larvae and pupae were primarily collected in streams acidic in nature but immatures also survived well in flows more neutral in pH reaction.

Simulium verecundum was found on dams or just downstream from impounded water at Shepard's Mill (Site 86), Chattahoochee (88), Tall Timbers (132) and below Lake Melrose (185). Other sites such as Hurricane Creek (83) and Site 90 could be traced to bodies of impounded water upstream. Although a few specimens were found at locations such as Holman Branch (82), Shaw Creek (91) and Camp Branch Creek (118) which were sandy and had little trailing vegetation (even here the larvae were concentrated on the thin sedges or few blades of grass or twigs in the flow), the largest populations were found where eel, other grasses, and additional large and small leafed aquatic vegetation were abundant. Larvae and pupae were found attached to grasses especially and to a wide

Figure 119. Panther Creek, Site 223, where *S. verecundum* was collected.

range of plants in the streams in addition to twigs and, on a few occasions, to the rock-like clay substrate of a stream. When construction at Sites 6 and 17 destroyed sections of the streams rich in vegetation and forced sampling upstream where the flows continued in a woods where aquatic vegetation was essentially lacking, *S. verecundum* no longer appeared in the collections. At Kettle Creek (130), the Waccasassa River and Site 196 *S. verecundum* was never or only rarely discovered in the main flows. Small sandy or sand-mud bottomed drainage ditches which emptied into the larger streams, however, harbored good numbers of *S. verecundum* on grass, sedges, dead leaves, shells and other substrates. The side flows differed from the main streams in size, velocity, temperature and at Kettle Creek the pH of the side flow was higher than the pH of the main flow.

Simulium verecundum was associated with 12 other species in Florida's streams (Table 3). Most frequently it was found with *S. tuberosum* and *S. slossonae*. On 30 occasions it was collected with *S. vittatum*. Only a few *S. verecundum* larvae with signs of protozoan infections have been collected and no mermithids have been noted in the immatures.

A character given in some taxonomic keys to separate *S. verecundum* larvae from its relative *S. venustum* is that *S. verecundum* possesses more than 52 rays in each cephalic fan. Few of the *S. verecundum* larvae collected in Florida have more than 52 rays and most range from 45-50. Also an unexpected fulvous area lines the gular notch on some specimens and others have an apparently uncharacteristic reddish abdominal tinge. The adults reared or dissected from immatures in populations with variant characters such as these have all proven to be *S. verecundum*.

Florida collection records for *S. verecundum*.

Alachua Co. 1) 1973: 7 Dec (S), 13 Dec (P); 1974: 2 Feb (M,L,P), 12 April (S,M), 5 May (P), 30 Oct (S,L), 23 Nov (M,L,P,A); 1975: 10 Jan (S,M), 12 Feb (S,L,P). 6) 1973: 17 Nov (S), 7 Dec (S,M); 1974: 2 Jan (M,P,A), 2 Feb (S,M,P), 7 March (S). 15) 1975: 31 Jan (P). 17) 1974: 2 Feb (S,M), 7 March (M,L,P), 12 April (S,M,L), 5 May (S,M,L,P), 6 June (S,M). 18) 1974: 25 Oct (M); 1975: 10 Jan (S), 31 Jan (S,L,P), 8 April (S,M,L), 11 May (S), 14 June (S). 32) 1974: 2 Feb (S,M,L,P,A), 9 March (S,P), 13 April (M,P,C,A); 1975: 10 Jan (S,M,L,P,A), 4 April (S,M,L,P,C). 39) 1974: 25 Oct (M).

Baker Co. 41) 1974: 4 Jan (S), 6 Nov (S).

Bradford Co. 42) 1975: 19 Nov (S). 47) 1975: 26 Jan (S,M,L), 26 April (S,M,P). 48) 1975: 26 Jan (P,A).

Dixie Co. 68) 1975: 17 Jan (S), 26 March (S,M,P,C,A).

Duval Co. 72) 1974: 20 April (S); 1975: 12 March (S,M,L,P,A), 4 May (S); 1976: 14 Feb (S).

Escambia Co. 73) 1974: 16 June (M); 1975: 12 June (S). 74) 1974: 16 June (S), 14 Oct (S,M,L,P); 1975: 19 Jan (S,L,P,A).

Gadsden Co. 81) 1973: 18 Dec (M); 1974: 17 March (S,M); 1975: 27 March (S,M,P,A). 82) 1975: 18 Jan (L). 83) 1974: 13 Oct (S); 1975: 18 Jan (S,M,L,P), 27 March (S,M,L). 86) 1975: 18 Jan (P,C,A). 87) 1974: 15 June (M,L,P); 1975: 18 Jan (S,M,L,P), 27 March (S,M,P), 29 Dec (S,L,P,A). 88) 1975: 30 Dec (P,A). 90) 1974: 17 March (S,M,L,P); 1975: 18 Jan (S,M,L,P,C,A), 27 March (S,M,L,P,C,A), 10 June (S), 29 Dec (S,M). 91) 1975: 27 March (P,A).

Hamilton Co. <u>101</u>) 1975: 1 Feb (S,M,L,P,A), 26 April (S).

Holmes Co. <u>118</u>) 1975: 18 Jan (S,M). <u>119</u>) 1975: 19 Jan (S,M).

Jefferson Co. <u>124</u>) 1975: 26 March (S).

Lafayette Co. <u>130</u>) 1975: 17 Jan (S,M,L,P,C,A), 26 March (S,M,L,P), 28 Dec (S).

Leon Co. <u>131</u>) 1974: 16 March (S,M,L); 1975: 26 March (S,M,L,P,C), 29 Dec (S). <u>132</u>) 1974: 16 March (S,M,P); 1975: 17 Jan (S).

Levy Co. <u>138</u>) 1975: 23 March (P). <u>139</u>) 1975: 23 March (S,M,P,C,A). <u>141</u>) 1975: 23 March (M).

Liberty Co. <u>142</u>) 1975: 18 Jan (S,M), 27 March (S,M). <u>147</u>) 1974: 17 March (S,M,P,A); 1975: 18 Jan (S), 27 March (S,M).

Madison Co. <u>153</u>) 1974: 16 March (S,M,P,C); 1975: 17 Jan (S,M,L,P), 26 March (S,M,L,P,C,A), 10 June (P), 29 Dec (S,M,P,C,A).

Nassau Co. <u>160</u>) 1975: 4 May (S). <u>161</u>) 1975: 12 March (S,P). <u>162</u>) 1975: 12 March (S,P).

Putnam Co. <u>185</u>) 1974: 26 March (S,M), 18 May (S,M); 1975: 15 Jan (S,M,P), 4 April (S,M,L,P,C), 11 May (S,M,L,P). <u>186</u>) 1974: 19 Jan (S), 14 April (M,L,P,A), 25 May (S,M,P), 23 Nov (M,P,C); 1975: 12 Feb (S,M,L,P,C,A), 18 April (S,M,L,P,C).

Santa Rosa Co. <u>196</u>) 1974: 18 March (S,M,L,P,A), 14 Oct (S,M); 1975: 28 March (S,M).

Seminole Co. <u>203</u>) 1975: 27 April (M).

Taylor Co. <u>210</u>) 1973: 17 Dec (S,M,L,P,C); 1974: 16 March (S,M,L,P); 1975: 17 Jan (S), 28 Dec (S,M,P,C). <u>213</u>) 1975: 17 Jan (S,M,L, P,C), 26 March (S,M,P,C,A). <u>215</u>) 1975: 26 March (S,M,P,C,A).

Union Co. <u>217</u>) 1974: 5 Jan (S); 1975: 12 Jan (L,P,A), 8 April (S).

Walton Co. <u>223</u>) 1975: 19 Jan (S,L,P), 28 March (M).

Cnephia species Undetermined No. 1

On 13 March 1969 in a small stream which passed under the Millhopper Rd. (State Road 232) .8 km (.5 mi) southeast of the Devil's Millhopper, Gainesville, Alachua County (Fig. 122), Mr. L. Goldman collected larvae of a black fly species which has been found nowhere else in Florida. The larvae with dark histoblasts are 6-7 mm long and appear pinkish brown in alcohol. The head spots are rather indistinct with only the two elongate, dark brown medial groups of spots very visible (Fig. 120). The antennae have dark brown basal segments, lighter brown second segments, and are very thin the rest of their lengths which extend beyond the cephalic fan stalk. The cephalic fans contain a large number of rays, about 80-90. The gular notch is a very tiny inverted-V (Fig. 121). The submentum has large broad lateral teeth and a shorter median tooth. The respiratory histoblast contains 20 to 30 long thin filaments which rise first in groups of 4 and 6 and then bifurcate in pairs off thicker, elongate stems. The abdomen in dorsal view bulges at the fifth segment, is widest at about the seventh segment, and tapers thereafter. There are about 64 rows of anal hooks with 10-12 hooks-row. The anal sclerite is elongate or tall with very short, dark, thin anterior and posterior arms. The anal tubercles are large.

The submentum of this species approaches that of *Cnephia mutata* (Malloch) and the size and shape of the throat cleft are somewhat similar to that of *C. mutata* but other characters disagree strongly such as the number of respiratory histoblast filaments and the shape of the anal sclerite.

The Goldman collection site in recent years has been disrupted by a housing development and no stream was observed to flow at the collection locality during this research.

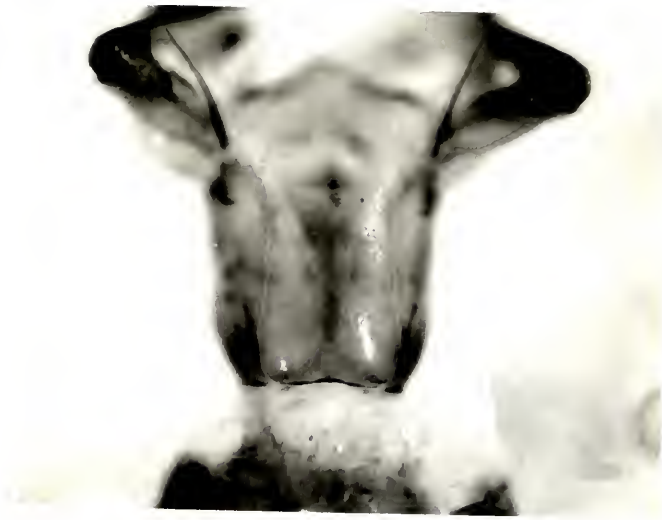

Figure 120. Cephalic apotome of a larva of *Cnephia species* No. 1.

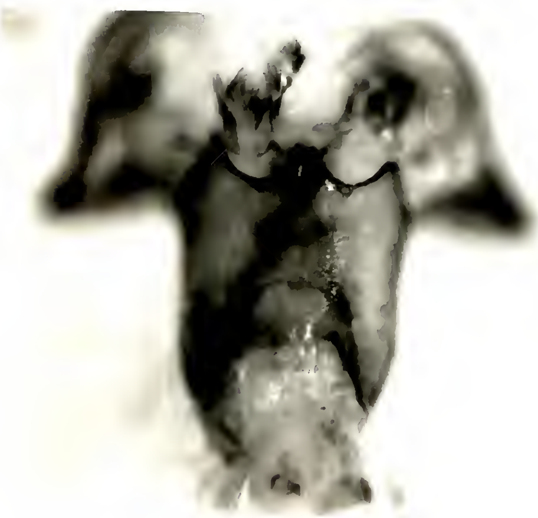

Figure 121. Venter of the head capsule of a larva of *Cnephia species* No. 1.

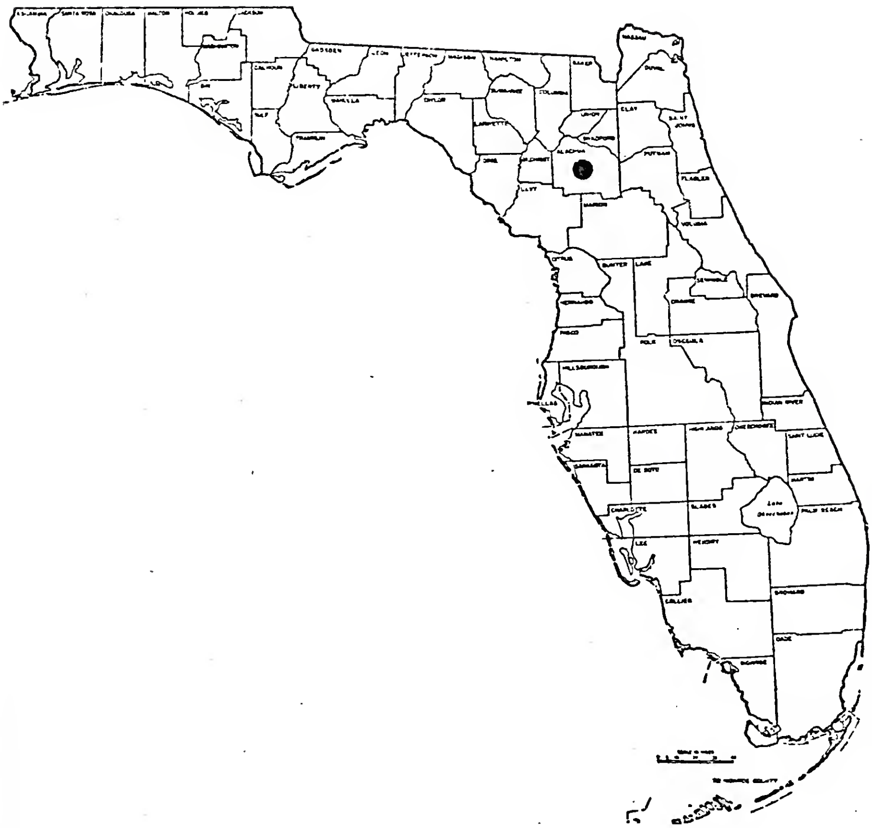

Figure 122. Collection location for *Cnephia species* No. 1 in Florida.

Simulium species Undetermined No. 1

On 19 January 1975 in the vicinity of Pine Barrens Creek (Site 74), Escambia County (Fig. 126), a pupal exuvium with cocoon was collected which is unlike any other black fly specimen found in Florida. The specimen, shown in Fig. 123, bears respiratory organs consisting of four filaments each rising in two pairs off short petioles. The long, thin filaments are about 3 mm in length. The cocoon is light brown, well-woven, has anterior edges which are concave in lateral view and bears a short mid-dorsal anterior projection. The cocoon is 3 mm long on the dorsum and 3.5 mm long ventrally.

The specimen was collected from pebbles or dead leaves in a small 25 cm wide, 2 cm deep side flow to Pine Barrens Creek (Fig. 127). The small stream emerged from a woods on a sand and round-pebble bed and flowed at .61 m/sec (2 ft/sec). The water temperature was 17.5°C (63.5°F) and the pH was 4.5. *Simulium tuberosum* larvae were also present in the small stream.

A medium-aged larva with incompletely-developed histoblasts was collected under the same conditions as the exuvium and is believed to represent the same species. The larva is 5 mm long and has a light brown head capsule and abdomen. The head spots appear dark brown on a lighter yellow brown cephalic apotome (Fig. 124). The posterior medial group of spots expands posteriorly. The antennae extend just beyond the stalk of the cephalic fan. The cephalic fans each consist of about 60 rays. The gular notch is longer than wide, rounded anteriorly and extends slightly less than halfway to the teeth of the submentum (Fig. 125). The submentum has prominent lateral teeth which are nearly as large as the conspicuous medial tooth. The anal sclerite has long anterior arms.

The anal tubercles are prominent.

The specimens appear to represent the subgenus *Eusimulium* and are similar but not identical to *S. clarkei* Stone and Snoddy, differing in the length of the filament petioles, and *S. latipes* (Meigen), which is a more northern species and has a long antero-dorsal projection on the cocoon. Attempts to collect additional specimens of this species during December 1975 and April 1976 were futile.

Figure 123. Pupal exuvium and cocoon of *Simulium species* No. 1.

Figure 124. Larval head spots of *Simulium species* No. 1.

Figure 125. Venter of the head capsule of the larva of *Simulium species* No. 1.

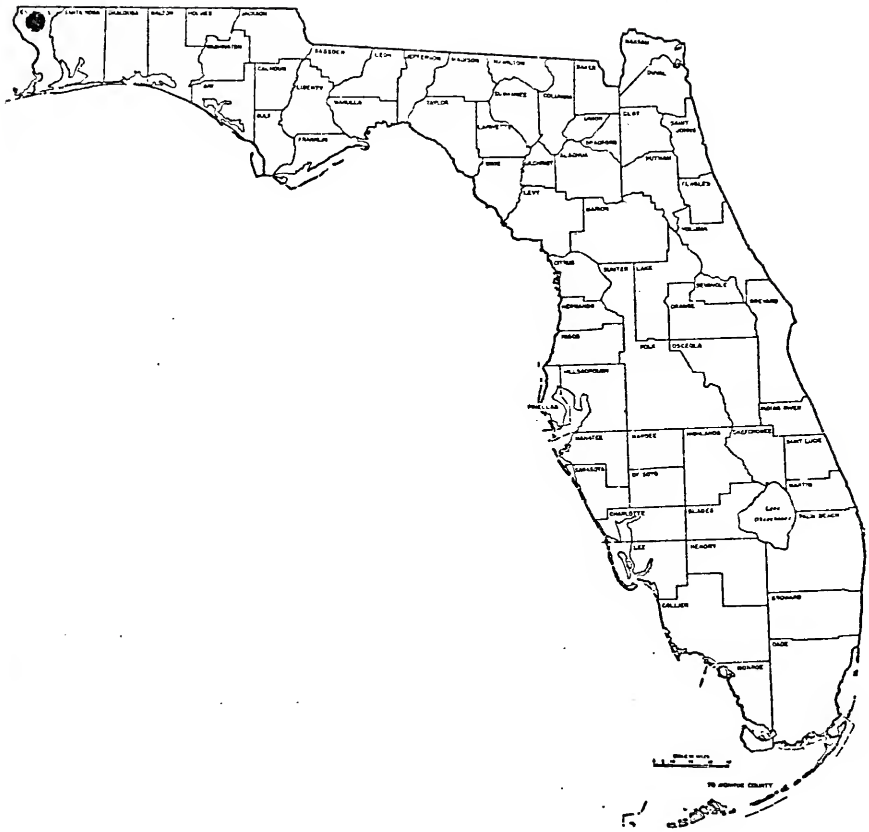

Figure 126. Collection location for *Simulium species* No. 1 in Florida.

Figure 127. Small flow to Pine Barrens Creek, Site 74, where *Simulium species* No. 1 was collected.

Leucocytozoon smithi Transmission

Table 5 presents the results from the sentinel turkey exposures. On four occasions one bird of each triplet exposed in Alachua County during May-July 1975 became infected with *L. smithi*. The presence of the disease and vectors indicated by the positive sentinels at the Hatchet Creek Preserve during May and July was further supported by blood smears positive for *L. smithi* which were obtained from two semi-wild turkeys (1/4 wild, 3/4 domestic) being reared in an outdoor pen at the Preserve. The first few positive sentinels (9170, 223 and 245) served as initial donors for lab transmission experiments. All subsequent lab transmission donors were lab-infected birds except during early 1976 when two sentinels, 483 and 485, were used as donors. Four turkeys infected during the summer and fall of 1975 were held over the winter to serve as donors for transmission trials early in 1976 if difficulties were encountered in 1976 as they had been in 1975 in acquiring younger, more manageable birds to field infect during January through March. It was hoped that an increase of the *Leucocytozoon smithi* infection reported in some *Leucocytozoon* species (Desser et al., 1967; Cook, 1971; Fallis et al., 1974) when the mating season began or under conditions of stress would occur in the overwintering laboratory birds. Mating usually begins during January or February with turkeys, and eggs were found frequently in the holding room beginning in February. Two of the female

Table 5 . Sentinel turkey locations and results.

Turkey Number	Exposure Unit	Location	Dates	*Leucocytozoon*
142, 143,	cage	Fisheating Creek (Site 95)	21-22 April 1975; 30-31 May 1975	negative
149, 150, 175	ramp trap	Lochloosa Creek (Site 22)	29-30 April 1975	negative
9168, 9169, 9172	ramp trap	SR 225/340 Junction (Site 8)	9-11 May 1975	negative
9163, 9167, 9170	cage	Hatchet Creek Preserve (Site 24)	17-23 May 1975	only 9170 positive, 2 June
245, 246, 247	cage	SR 225/340 Junction	16-22 June 1975	only 245 positive, 11 July
223, 224, 312	cage	Lochloosa Creek	2-10 July 1975	only 223 positive, 20 July
313, 314, 315	ramp trap	Hatchet Creek Preserve	3-10 July 1975	only 315 positive, 20 July
486, 487, 488	cage	Hatchet Creek Preserve	18-25 Feb 1976	negative
479, 480, 481	cage	Lochloosa Creek	4-8 March 1976	(birds stolen)
476, 477, 478	cage	SR 225/340 Junction	4-8 March 1976	(birds stolen)
483, 484, 485	cage	SR 225/340 Junction	9-16 March 1976	483, 484, 485 all positive, 22 March

birds (223 and 325) were severely wounded on the posterior lateral regions of their bodies during the mating attempts of a huge tom turkey initially housed with the females. Despite the flow of sex hormones and the stress of egg laying, mating and mating wounds, and very cold nights spent with little significant artificial heat, as of early March no significant rise in the gametocytemias of the birds was observed and they were sacrificed. One bird (324) previously exposed in the field on 7 January and fed upon by several black flies had a noticeable but short-lived rise in its gametocyte count approximately two weeks after the field exposure.

Temperatures during the day from 10-15°C (50°F) and at night around 0°C (28-35°F) which required that the sentinel poults be brought in at night might explain the negative results in the 18-25 February 1976 exposure. Although turkeys and equipment were stolen from the sentinel sites during the important first week in March 1976 when spring species such as *S. congareenarum* and *C. ornithophilia* were flying, the three heavy infections achieved in sentinels the following week compensated for the losses. One of these sentinels (483) with a heavy gametocyte level of 1-2 gametocytes per oil immersion field was the only turkey observed to die from other than preplanned causes. It cannot be stated for certain that *L. smithi* was the reason for its demise. Positive infections were obtained in this research in sentinels exposed primarily on the ground, on a high stream bank (Fig. 2) or on a platform 1.53 m above the ground. Observation of the efforts of Dr. D.J. Forrester in 1974 at Fisheating Creek revealed that sentinel birds placed in cages in cypress trees 6 m (20 ft) above the ground and exposed for two week periods during the summer became positive for *L. smithi* but poults

placed in similar cages at ground level remained negative for the disease.

Results from the Manitoba trapping were presented in Table 4 and the results from the ramp, blackout box, and exposed turkey collecting are presented in Tables 6, 7 and 8. The Manitoba captures revealed that a number of species were on the wing including species in the *Phosterodoros* group, for example at Lochloosa Creek where immature populations of a number of the *Phosterodoros* species occurred. The ramp trap and blackout box collections revealed the presence of only a single species, *S. slossonae*. The blackout traps were used on days when temperatures ranged from 25-32.2°C (77-90°F) and relative humidities ranged from 58-90% with the higher humidities most often encountered. Each successful trapping period yielded 1 to 16 specimens of *S. slossonae* and when an infected host was used as the attractant, blood-fed flies which were collected provided a starting point for early lab transmission trials. While the birds were uncovered in the blackout box trials it was noted that they were extremely sensitive to the presence of even a single black fly. They reacted by shaking, rubbing or pecking, which prevented many flies from feeding and killed numerous flies. These difficulties might have been overcome by restraining the host in some manner, but it was found that even the most docile restrained birds became very agitated and, if on their sides, struggled vigorously to stand to move their bowels or for some other reason. It was also observed that a number of black flies landed and fed on the head, neck or legs of the host. Some crawled beneath the feathers and emerged a few minutes later blood red and quickly flew off while the turkey was still in the uncovered condition. The ramp trap was hoped to be an improvement

Table 6 . Black flies captured in ramp traps.

Turkey Number	Location	Dates (all 1975)	Captures (all females)
149, 150, 175	Lochloosa Creek (Site 22)	29-30 April	0
9172, 9168, 9169	Rt 225/340 Junction (Site 8)	9-11 May	0
9170	Hatchet Creek Preserve (Site 24)	18-20 June	0
9170	Rt 225/340 Junction	21-22 June	1 *S. slossonae*
313, 314, 315	Hatchet Creek Preserve	3-10 July	2 *S. slossonae*

Table 7 . Blackout box trapping results.

Turkey Number	Location	Date	Time	Captures (all females)
121	Lochloosa Creek (Site 22)	15 Aug 1974	1730-1845	*S. slossonae*
121	Lochloosa Creek	20 Aug	1755-1930	*S. slossonae*
121	Lochloosa Creek	27 Aug	1800-1930	*S. slossonae*
121	Lochloosa Creek	28 Aug	0730-0930	*S. slossonae*
121	Lochloosa Creek	12 Sept	1620-1945	*S. slossonae*
121	Tributary to Lochloosa Creek (Site 23)	25 Sept	1550-1900	*S. slossonae*
121	Stream from Lake Melrose (Site 185)	14 May 1975	1430-1830	0
121	SR 225/340 Junction (Site 8)	3 June	0900-1130	*S. slossonae*
121	Sandy Hatchet Creek (Site 1)	3 June	1140-1230	*S. slossonae*
9170	Flow to Newnan's Lake (Site 18)	3 June	0830-0930	0
9170	Hatchet Creek (Site 20)	3 June	1000-1215	0
9170	Turkey Creek (Site 216)	15 June	1740-2020	*S. slossonae*
223, 224, 225	Lochloosa Creek	2 July	1700-1730	0
9170	Double Run Creek (Site 43)	6 July	0830-0940	*S. slossonae*
9967, 9947	Fisheating Creek (Site 95)	18 Sept	0900-1130	0

Table 8. Black fly captures from exposed turkeys.

Turkey Number	Exposure Location	Date	Time	Captures
142, 143	Fisheating Creek (Site 95)	21 April 1975	1600-1700	0
142, 143	Fisheating Creek	22 April	1200-1500	0
313, 314, 315	Hatchet Creek Preserve (Site 24)	5 July	1700-1800	*S. slossonae*
9170	Double Run Creek (Site 43)	6 July	0945-1110	*S. slossonae*
9170, 245	Sandy Hatchet Creek (Site 1)	19 July	0830-1100	*S. slossonae*
223	Sandy Hatchet Creek	22 July	0830-1030	*S. slossonae*
9947, 9967 9969	Fisheating Creek	29 July	0930-1100	0
316, 317	Lochloosa Creek (Site 22)	5 Aug	1030-1225	*S. slossonae*
320, 9966	Lochloosa Creek	30 Aug	1000-1200	0
320, 9966	SR 225/340 Junction (Site 8)	30 Aug	1740-1930	*S. slossonae*
324	Double Run Creek	7 Jan 1976	1330-1510	*S. slossonae*
324	Turkey Creek (Site 216)	7 Jan	1600-1715	*S. congareenarum*, *S. slossonae*
479, 480, 481	Lochloosa Creek	4 March	1600-1630	*S. congareenarum*, *S. slossonae*
476, 477, 478	SR 225/340 Junction	4 March	1500-1530	*S. congareenarum*, *S. slossonae*
485	Double Run Creek	24 March	1400-1600	*S. slossonae*
485	Turkey Creek	24 March	1645-1830	0
483	Sante Fe College (Site 28)	26 March	0930-1030	0
485	SR 225/340 Junction	31 March	1600-1730	*S. slossonae*

in that it still used turkeys as attractants; however, flies entering to feed were considered captured and the collector could recover specimens as his schedule allowed. The ramp trap used in Alachua Co. and the 4 to 5 ramp traps used in Glades Co., primarily in another study for five 2-week periods during May-October 1975, captured many mosquitoes and other insects but only three black flies were recovered in Alachua Co. and none in Glades Co. The black flies captured, being the ornithophilic species *S. slossonae*, were significant and gained in importance when one of the sentinel birds (315) used during the 3-10 July 1975 exposure period when two *S. slossonae* were recovered from the trap developed leucocytozoonosis. However the success of capturing simuliids with ramp traps overall was poor. The possible inappropriateness of the ramp trap for capturing black flies was suggested during a little experiment in June 1975 that was conducted behind the Veterinary Entomology lab. At the slanted entrance slit of the ramp trap four reared *S. jonesi* females were released. Three of the four flies released flew to the upper screened portion of the trap opposite from the entrance slit where the release occurred. One black fly flew a short loop out of the vial into the trap and landed halfway down the slanted ingress screen, clinging upside down. It immediately commenced the typical black fly walking and wandering behavior observed often in the collecting jars of the Manitoba traps. The black fly walked up the slanted entrance ramp on the inside to the metal frame securing the organza and immediately flew out the narrow 2.5 cm (1 in) entrance slit and escaped. Downe and Morrison (1957) reported that *S. vittatum* and *S. decorum* readily entered animal baited traps through baffle entries but mentioned, once the flies were inside, they ignored the host and searched for a means to escape.

Perhaps this behavior plus reduced visibility of target animals or restricted flow of host attractant odors through the wood and fine-meshed polyester organza detracted from the effectiveness of the trap.

The flies captured from the exposed turkeys reveal the presence during January and March of a second ornithophilic species, *S. congareenarum*, which was observed to feed on the turkeys. Black flies approached the turkeys on days when the temperatures ranged from 22.2°C (72°F) to 30.6°C (87°F) and the wind was calm or blew in gusts up to 16 km/hr. Rarely were situations encountered where more than two or three flies of any species were attracted to or were observed feeding on one turkey at the same time. Circumstances were never experienced such as Garris et al. (1975) report where several hundred black flies were observed around a single turkey in one half hour period. It was noted that, while black flies were in flight and were collected from exposed turkeys during almost all periods during the day, the number of simuliids around exposed turkeys and around my head increased with the approach of thunderstorms during the summer afternoons.

Black flies were observed to bite and feed on turkeys in the field and in the lab at a number of body regions. Biting was confirmed when a stationary fly in the head-down feeding position was observed to pull its mouthparts out of the skin and blood feeding was noted by a hematoma on the skin and bright red blood in the gut of the dissected fly. In the field flies were observed to crawl beneath the feathers of the wing and the lower abdominal region to feed and also fed on the top of the head, on the neck, and on bare portions of the legs. In the lab positive feedings were achieved by placing flies against the breast and bare underside of the wing as well as on the neck and head. Compared to the

numbers of flies of other species that fed on turkeys in the field or in the lab many more *S. slossonae* were observed to feed on the birds. Both wild-caught and reared specimens of *S. slossonae* and *S. congareenarum* fed on turkeys. Reared *S. congareenarum* females were very reluctant to feed on the turkeys, regardless of whether they were young or old flies, were provided with a sugar source or deprived of food for 24-48 hours, were provided with water or deprived of water for 3-12 hours, held with males or held alone or even taken into the field in hopes of stimulating feeding. Reared, non-blood fed *S. congareenarum* dissected during January 1976 were discovered to have large, well-developed eggs which suggests at least some autogenous egg development. Six reared *C. ornithophilia* fed on the turkeys as did six wild-caught *S. meridionale*. No *S. meridionale* adults were reared. In addition, reared females of the *S. (Phosterodoros)* species *jenningsi*, *jonesi*, and *notiale* and undetermined wild-caught *S. (Phosterodoros)* species fed on the turkeys. *Simulium tuberosum* would not feed on turkeys in the lab. In the lab flies fed on turkeys during all hours of the day, especially during the late afternoon, and even when it was dark outside. Black flies stimulated to feed usually went right to the turkey when the feeding vial was placed against the bird and were biting within five minutes. Only rarely was feeding stimulated when flies reluctant to feed were kept on the turkeys thirty minutes or longer. Garris et al. (1975) observed black flies feed to such repletion on turkeys that they could not fly and would fall to the ground. During two years of feeding black flies on turkeys under many different circumstances I have noted that once the black flies become replete with blood they remove their mouth parts and always quickly fly away if unrestrained. If confined in a feeding vial

they exhibit for a short period a burst of flight activity after which they settle down and become quiescent for a number of hours.

Simulium slossonae was observed to feed under a variety of temperatures (13 -32°C = 55 - 90°F) and relative humitidies (40-90%). Uninterrupted feeding times for *S. slossonae* ranged from 1 to 10 minutes with an average of 4.3 minutes. *Simulium congareenarum* fed at temperatures from 17-27°C (62-80.5°F) and at relative humitidies of 60-87%. *Simulium congareenarum* feeding times ranged from 1.5 to 5 minutes with most being 3-5 minutes. On a number of occasions in the field and lab a small droplet of blood was observed to protrude from the anus of *S. slossonae* just prior to terminating feeding. Most black flies fed to repletion rapidly, often in three to four minutes, and were not readily disturbed while engorging. Complete engorgement occurred most swiftly, in as little time as one minute, when flies fed on the posterior portion of the head or on the neck. Longer feeding periods were apparently spent attempting to create a pool of blood beneath the skin. Actual engorgement marked by a swift ballooning of the fly's abdomen as the gut filled with blood occurred rapidly, in about one minute. One *S. congareenarum* aspirated out of a Manitoba trap in the field was placed against a turkey's neck while still in the aspirator tube. The fly walked to the small neck feathers and clinging to the top side of a feather with only the proboscis touching the skin of the turkey proceeded to engorge in three minutes (1729-1732 hrs). *Cnephia ornithophilia*, a large fly, took a larger blood meal and required a longer feeding period, average 8.5 minutes, than usual for the other species observed. One *Cnephia* female propped itself up in the typical feeding position for this species which is at a 75 to 80° angle almost

perpendicular to the host's body surface, inserted the mouth parts at 1550 hrs and remained attached, not filling with blood and releasing until 1606 hrs. The hematomas created by feeding *Cnephia* on the chest of a turkey, often 3 mm in diameter, were usually larger than the feeding marks caused by other species. On 25 March a 5 mm hematoma resulting from a 4 minute feeding by *S. congareenarum* was noted. *Simulium meridionale* fed within 2 to 8 minutes and seemed to engorge more extensively than any other species with the abdomen expanding to about three times the size of the thorax.

Table 9 presents a summary of the successful transmission trials. Three black flies, *S. slossonae*, *S. congareenarum* and *S. meridionale*, all proven as vectors in other States, were found to be pathophorous agents of *L. smithi* in turkeys in Florida. Of 116 *S. slossonae* that fed once on turkeys, 26 fed at least twice - once on an infected bird and one or more times on clean turkeys - and 15 positive transmissions occurred. Most transmissions were achieved by the bite of a single fly. Two wild caught *S. congareenarum* fed directly on a clean turkey with no leucocytozoonosis resulting. Of 7 *S. congareenarum* that fed once on infected birds, 3 of the 4 that survived fed a second time, on clean birds, and 3 transmissions occurred. With *S. meridionale* 6 females fed one time on infected turkeys, 1 fly fed for a second time and *L. smithi* transmission was achieved. Injections of possibly infected flies ground and innoculated in physiological saline intraperitoneally into turkeys were tried on four occasions with *S. slossonae* and on one occasion with *S. meridionale* with negative results. Two females of *S. notiale* and one of *S. taxodium* fed once on infected hosts but died soon thereafter or would not feed again on uninfected hosts. One *S. jenningsi* female, one

Table 9. *Leucocytozoon smithi* transmissions.

Case No.	Species	Vector Source	Donor	Vector Infected	Recipient	Transmission Date	Positive Smear
1	*S. slossonae*	field exposure	9170	6 July 1975	316	13 July	23 July
2	*S. slossonae*	Manitoba trap	9170	9 July 1975	317	13 July	27 July
3	*S. slossonae*	field exposure	223	22 July 1975	320	27 July	11 Aug
4	*S. slossonae*	reared	316	6 Aug 1975	9966	10 Aug	21 Aug
5	*S. slossonae*	Manitoba trap	316	10 Aug 1975	224	15 Aug	28 Aug
6	*S. slossonae*	field exposure	9966	30 Aug 1975	321	2 Sept	12 Sept
7	*S. slossonae*	Manitoba trap	321	19 Sept 1975	339	23 Sept	5 Oct
8	*S. slossonae*	reared	321	30 Sept 1975	340	2 Oct	17 Oct
9	*S. slossonae*	reared	321	30 Sept 1975	342	3 Oct	17 Oct
10	*S. slossonae*	reared	321	2 Oct 1975	325	6 Oct	17 Oct
11	*S. slossonae*	reared	342	21 Nov 1975	324	25 Nov	6 Dec
12	*S. congareenarum*	field exposure	324	7 Jan 1976	9056	11 Jan	24 Jan
13	*S. slossonae*	field exposure	485	24 March 1976	412	27 March	8 April
14	*S. congareenarum*	Manitoba trap	485	25 March 1976	411	28 March	8 April
15	*S. slossonae*	Manitoba trap	485	25 March 1976	416	29 March	8 April
16	*S. congareenarum*	field exposure	485	24 March 1976	404	27 March	11 April
17	*S. slossonae*	field exposure	485	31 March 1976	415	3 April	15 April
18	*S. meridionale*	netted	411	19 April 1976	414	22 April	4 May
19	*S. slossonae*	netted	411	19 April 1976	417	22 April	7 May

S. jonesi, and three *C. ornithophilia* fed twice but no *L. smithi* infections resulted.

The vectors listed in Table 9 were collected from infected turkeys exposed in the field (field exposure), captured in Manitoba traps, were reared, or were netted. The collection locations of the vectors include 10 different sites in a total of 7 west, north, central and south Florida counties. The black flies were experimentally infected during nearly every month of the year with most transmissions occurring during late summer-early Fall (August-early October) and in the Spring (March-April). Flies held for 2-7 days after taking an infected blood meal transmitted the disease. Transmissions were achieved with feeding times that ranged from about 1 minute in interrupted feedings to 10 minutes for some full engorgements. One reared *S. slossonae* infected on 30 September transmitted *L. smithi* to host #340 on 2 October in an interrupted feeding and the same fly transmitted *L. smithi* to turkey #342 the next day, 3 October, during a more complete feeding. Recipients for all transmissions were young turkeys from 18 days to 19 weeks old, with most being 3-7 weeks of age. The prepatent period from the transmission date when an infected fly bit the clean turkey to the appearance of gametocytes (Fig. 128) in the blood smears was 10-15 days.

Deeply staining ookinetes of *L. smithi* (Fig. 129) 20-22 μ long and 2.5-3.5 μ wide with large crystalloid vacuoles have been noted in smears of the gut contents of *S. slossonae* 11 hours after a blood meal was taken from an infected turkey and a little later in the gut contents of infected *S. congareenarum*. Spherical hyaline structures on the midgut believed to be oocysts were observed many times in flies that had fed on infected turkeys but unruptured structures displaying distinct

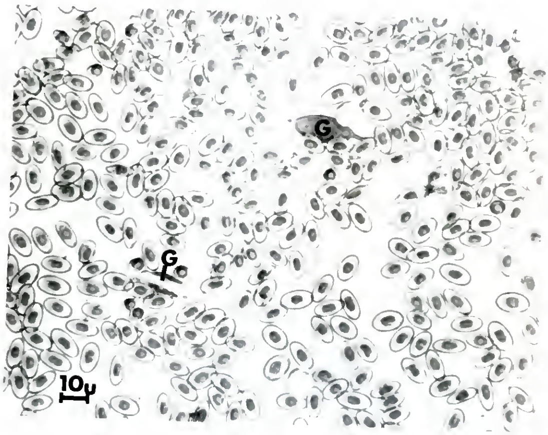

Figure 128. Gametocytes (G) of *L. smithi* among normal turkey blood cells.

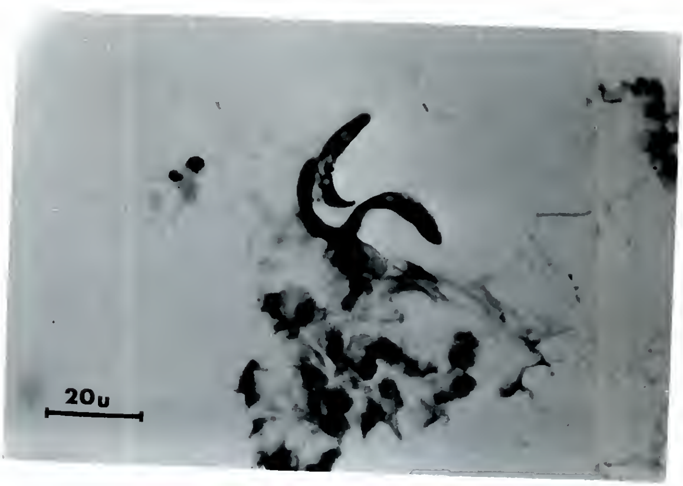

Figure 129. Ookinetes of *L. smithi*.

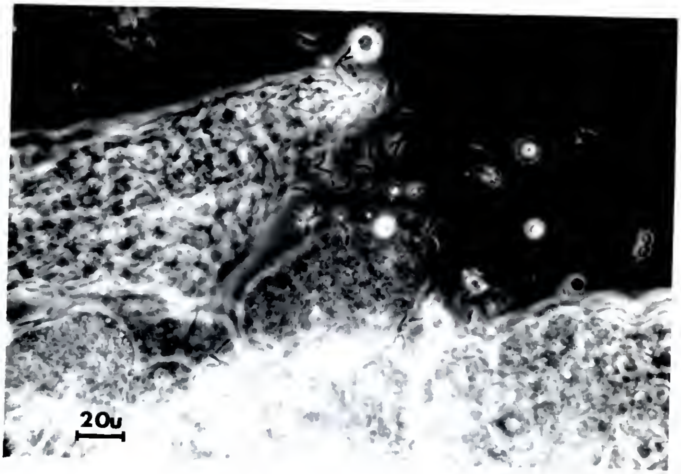

Figure 130. Sporozoites of *L. smithi* photographed in saline.

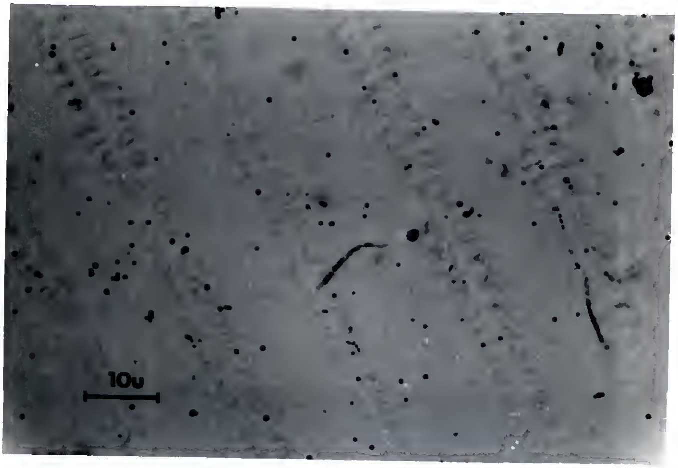

Figure 131. Stained sporozoites of *L. smithi*.

elongate sporozoites packed like toothpicks as illustrated for other species of *Leucocytozoon* (Fallis and Bennett, 1961a) were never positively located. Sporozoites (Fig. 130 and 131) about 10-15 μ long and 1.25-2 μ wide were observed issuing in a saline preparation from the gut region, probably from squashed oocysts, 45 hours after the female *S. slossonae* fed on a heavily infected turkey. In a *S. slossonae* female that had taken an infected blood meal 72 hours prior to dissection 50-100 sporozoites were observed escaping into the saline from the salivary glands and only a few sporozoites were noted in the gut region. The sporozoite stage with a blunt or rounded and wider anterior end and a tapering posterior end appeared slightly longer and wider in a fresh saline preparation in contrast to the fixed and stained preparations. Sporozoites located on the slides of guts and salivary glands of flies which transmitted *L. smithi* to turkeys were normally few in number except in one instance when the vector was allowed to insert its proboscis for only 45 seconds. The salivary glands of this fly were found to contain many sporozoites.

From the above investigations it is concluded that *Simulium slossonae*, the most widely distributed species of the three vectors and the only species present year round is the most important vector of *L. smithi* to turkeys in Florida. *Simulium congareenarum* is a secondary vector of significance especially during the first three or four months of each year. In the counties bordering the Apalachicola River, during the two to three week mass emergence period of *S. meridionale* in April and during most of May when the adults are still flying, *S. meridionale* may be an important vector of *L. smithi*.

CHAPTER V
CONCLUSIONS

1) From this research it is concluded that sixteen previously described species representing two genera and six subgenera of black flies occur in Florida. In addition, two species not positively identified and possibly previously undescribed are recorded from the State. Eight of the known species and the two undetermined species are reported for the first time from Florida.

2) Species such as *S. slossonae* and *S. tuberosum* are widely distributed throughout much of the State, are adapted to a wide range of lotic habitats and are present all year. Certain species such as *S. dixiense*, *S. haysi*, and *S. notiale* have a limited distribution in Florida and occur at only a few locations. Other species such as *C. ornithophilia*, *S. congareenarum*, and *S. verecundum* were found to have a more limited seasonal occurrence and were undetected during the warm summer months.

3) Except during mass emergences of *S. meridionale* in April along the Apalachicola River, black flies rarely cause harm directly to man in Florida. The potential for the transmission of diseases, especially viral encephalitis, by simuliids nevertheless exists.

4) Three species of black flies already recognized as vectors in other areas of the U.S. were incriminated through lab and field efforts

as vectors of *L. smithi* to turkeys in Florida. In order of decreasing importance, these species are: *S. slossonae*, *S. congareenarum*, and *S. meridionale*.

5) Barring major breakthroughs in prophylatic medications or vector control, the widespread distribution of the ornithophilic vectors of *L. smithi* would restrict any potential turkey industry to locations miles away from black fly streams or force screened outdoor or indoor rearing procedures.

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APPENDIX
COLLECTION SITE NAMES AND LOCATIONS

Alachua Co.

1) Sandy Hatchet Creek. 3.4 km north of Highway 24 on State Road (SR) 225. R 20 E-T 9 S, Section 2, SE quadrant.

2) Blues Creek. 1.6 km north of SR 232 at the IFAS Horticulture Unit. R 19 E-T 9 S, Section 9, SW quadrant.

3) Devil's Millhopper. 2.4 km north of SR 222 on SR 232. R 19 E-T 9 S, Section 15, SE quadrant.

4) Tributary to Hogtown Creek. .8 km south of the Devil's Millhopper on SR 232. R 19 E-T 9 S, on the border of Sections 22 and 23.

5) Gainesville. (Adult black fly captured biting a human -- D. Young.)

6) Tributary of Hatchet Creek. 5.6 km northeast of the SR 225/Highway 24 junction, on Highway 24. R 21 E-T 9 S, Section 6, NE quadrant.

7) Little Hatchet Creek. 1.6 km northeast of the SR 232/Highway 24 junction, on Highway 24. R 20 E-T 9 S, Section 23, NE quadrant.

8) Tributary to the Santa Fe River. .3 km north of the SR 225/340 junction, on SR 225. R 20 E-T 8 S, Section 15, NE quadrant.

9) Tributary to the Santa Fe River. 1.3 km northeast of the SR 225/340 junction, off SR 225 in the woods. R 20 E-T 9 S, Section 14, NW quadrant.

10) Tributary to the Santa Fe River. 1 km west of the SR 225/340 junction, on SR 340. R 20 E-T 8 S, Section 15, NW quadrant.

11) Stream to Newnan's Lake. 1.2 km north of SR 225/Highway 20 junction, on SR 225. R 20 E-T 10 S, on the border of Sections 1 and 2.

12) At a cypress dome. 3.2 km northwest of Gainesville off Route 441. (Adult black flies captured from chickens -- H. Davis.)

13) Gainesville, Univ. Florida campus. Northeast about 20 m from the intersection of Museum Road and Center Drive. R 20 E-T 10 S.

14) Tributary to the Santa Fe River. 1.6 km west of SR 225. R 20 E-T 8 S, center of Section 27.

15) Hogtown Creek. Gainesville at the junction of University Avenue and 34th Street. R 19 E-T 10 S.

16) Rattlesnake Branch. Gainesville. At the north end of NW 20th Terrace off 8th Avenue. R 20 E-T 10 S.

17) Hatchet Creek. 4 km north of the SR 225/Highway 24 junction, on Highway 24. R 21 E-T 9 S, Section 7, NW quadrant.

18) Flow to Newnan's Lake. 4.2 km southwest of the SR 232/Highway 26 junction, on Highway 26. R 20 E-T 10 S, Section 31, SW quadrant.

19) Santa Fe River. SR 225 at Alachua-Bradford County border. R 21 E-T 7 S, Section 32, NE quadrant.

19a) Santa Fe River. SR 235 at Alachua-Bradford County border. R 20 E-T 7 S, Section 18, SE quadrant (Stone and Snoddy, 1969).

20) Hatchet Creek. On Highway 26 1.6 km east of the junction of SR 232/Highway 26. R 21 E-T 9 S, Section 21, SE quadrant.

21) Lochloosa Creek. On Highway 20 6.4 km east of SR 234/Highway 20 junction. R 22 E-T 10 S, Section 30, NE quadrant.

22) Lochloosa Creek. 6.4 km east of SR 325 and 1.6 km north of Sike's Fish Camp. R 22 E-T 11 S, Section 6, NE quadrant.

23) Tributary to Lochloosa Creek. 4.8 km east of SR 325 and .5 km north of Sike's Fish Camp.

.24) Hatchet Creek. Off Highway 26 1.8 km east of the junction of SR 232/Highway 26, .6 km north of the highway in the woods at the Hatchet Creek Preserve. R 21 E-T 9 S, Section 22, SW quadrant.

25) Austin Cary Memorial Forest. 8 km northeast of the junction of SR 225/Highway 24, up Highway 24. Adult black flies captured in a Malaise trap by J. Glick.

26) Tributary to Hogtown Creek. Gainesville, in the woods east of 34th Street and NW 34th Ave. R 19 E-T 9 S, Section 26, SE quadrant.

27) Tributary to Hogtown Creek. Gainesville, in the woods east of 34th Street and NW 27th Avenue. R 19 E-T 9 S, Section 26, SE quadrant.

28) Gainesville, in the woods south of NW 23rd Avenue and NW 83rd Street at Sante Fe College. R 19 E-T 9 S, Section 32 NE quadrant.

29) Hogtown Creek. Gainesville, on SR 232A 4 km southeast of Highway 23 at the Gaineswood Apartments. R 20 E-T 9 S.

32) Hogtown Creek. Gainesville, on SW 20th Avenue 3.0 km west of 34th Street. R 19 E-T 10 S, Section 10, SE quadrant.

33) Gainesville, Univ. Florida campus. Stream from Graham and Simpson Hall ponds at intersection of North-South Drive and Museum Road. R 20 E-T 10 S.

34) Gainesville, Univ. Florida campus. Stream near Veterinary Entomology Building at the bottom of the hill on Newell Drive. R 20 E-T 10 S.

36) Headwaters of Hatchet Creek. Off SR 225 .8 km due west, across from NE 73 Avenue. R 20 E-T 9 S, Section 11, SW quadrant.

38) Lacrosse, Jamnback and Stone, 1957.

39) Gainesville, at the spillway just east of the Patuxent Wildlife Research Center on Highway 26 1.2 km west of SR 225. R 20 E-T 10 S, Section 2, west center.

Baker Co.

41) Turkey Creek. 1.6 km south of Route 90 on SR 121 at MacClenny. R 22 E-T 3 S, Section 5, NE quadrant.

Bay Co.

40) Juniper Creek. 4.8 km north of Highway 20 on Route 231. R 12 W-T 1 N, Section 14, west center.

Bradford Co.

42) Prevatt Creek. Near Starke, 5.6 km north of the junction of Highway 18 and Route 301, on Route 301. R 22 E-T 7 S, Section 6, SE quadrant.

43) Double Run Creek. 4 km southeast of Hampton on Highway 18. R 22 E-T 7 S, Section 27, SW quadrant.

44) Water Oak Creek. 3.5 km north of the Highway 16/Route 301 junction, on Route 301, north of Starke. R 22 E-T 6 S, Section 10, SW quadrant.

45) Sampson Creek. 8 km west of the Highway 18/Route 301 junction, on Highway 18, west of Graham. R 21 E-T 7 S, Section 29, NW quadrant.

46) Drainage flow to Double Run Creek. 4 km southeast of Hampton on Highway 18. R 22 E-T 7 S, Section 27, SW quadrant.

47) Olustee Creek. In Lawtey .4 km north of the SR 225/Route 301 junction, on Route 301. R 22 E-T 5 S, Section 23, SE quadrant.

48) Small flow west of Brooker on Highway 18 at the junction with SR 237. R 19 E-T 7 S, Section 1, SE quadrant.

49) Juniper Creek. 9.7 km west of Clarkesville on Highway 20. R 11 W-T 1 S, Section 1, west center.

50) Chipola River. 2.4 km west of SR 275 on Highway 20. R 9 W-T 1 S, Section 6, NE quadrant.

51) Apalachicola River. 3.0 km east of Blountstown on Highway 20. R 8 W-T 1 N, Section 35, NE quadrant.

52) Four Mile Creek. 1.4 km north of the Highway 73/Highway 20 junction, on Highway 73. R 9 W-T 1 N, Section 31, NW quadrant.

53) Blountstown. J. Yelvington collected *S. meridionale* adults on 10 May 1973 from young chickens. R 8 W-T 1 N.

Clay Co.

55) Gold Head Branch. At Gold Head Branch State Park 9.7 km northeast of SR 100/Highway 21 junction on Highway 21. R 23 E-T 8 S, Section 1, NE quadrant.

56) Tributary to Ates Creek. 8.9 km northeast of Gold Head Branch State Park on Highway 21. R 24 E-T 7 S, Section 3, SW quadrant.

57) South Prong Black Creek. 4 km south of Highway 16 on Highway 21. R 24 E-T 6 S, Section 27, SW quadrant.

Columbia Co.

58) Santa Fe River. At Oleno State Park 3.2 km east of Route 441. R 17 E-T 7 S, Section 2, NE quadrant.

59) Ichetucknee River. On Route 27 at the Columbia/Suwanee County border. R 15 E-T 6 S, Section 23, NW quadrant.

60) Santa Fe River. On Route 27 at the Alachua/Columbia County border north of High Springs. R 17 E-T 7 S, Section 32, NE quadrant. (Young, undetermined *S. (Phosterodoros) spp.* larvae collected.)

61) Santa Fe River. 2.4 km south of SR 138 at the Gilchrist/Columbia County line. R 16 E-T 7 S, Section 36, SW quadrant. (Young, undetermined *S. (Phosterodoros) spp.* larvae collected.)

62) Santa Fe River. 2.4 km south of SR 138 at the Gilchrist/Columbia County line, side flow in the woods. R 16 E-T 7 S, Section 36, SW quadrant.

Dade Co.

63) Biscayne Bay, Florida. Mrs. A.T. Slosson, black fly adults.

Desoto Co.

64) Prairie Creek. 2.4 km northwest of the Charlotte/Desoto County line on Highway 31. R 25 E-T 39 S, Section 26, SE quadrant.

65) Joshua Creek. On Highway 31 .2 km north of the Highway 31/SR 760 junction, southeast of Arcadia. R 25 E-T 38 S, Section 16, NW quadrant.

66) Buzzard Roost Branch. On Highway 70 13 km northwest of Arcadia. R 23 E-T 37 S, Section 10, SW quadrant.

Dixie Co.

67) Tributary to the Steinhatchee River. 23 km northwest of Cross City on Route 98. R 10 E-T 8 S, Section 33, NE quadrant.

68) On Route 98 within 1.6 km northwest of Cross City. R 12 E-T 10 S, Section 5, SE quadrant.

69) Pumpkin Swamp Creek. 13 km northwest of Cross City on Route 98. R 11 E-T 9 S, Section 20, SW quadrant.

Duval Co.

70) Yellow Water Creek. 9.7 km northeast of SR 228/Route 301 junction on SR 228. R 24 E-T 3 S, Section 20, NE quadrant.

71) Jacksonville. Mrs. A.T. Slosson, black fly adult.

72) 6.4 km west of the SR 115c/Route 90 junction on Route 90. R 24 E-T 2 S, Section 20, SE quadrant.

Escambia Co.

73) Canoe Creek. 13.7 km into Escambia Co. going west on Highway 4. R 31 W-T 5 N, Section 8, NW quadrant.

74) Pine Barrens Creek. 4.8 km south of the Highway 4/Highway 99 junction, on Highway 99. R 32 W-T 5 N, Section 28, NE quadrant.

75) McDavid Creek. 17.7 km south of the Highway 97 and Highway 99 junction, on Highway 99. R 32 W-T 3 N, Section 33, NE quadrant.

Flagler Co.

77) Little Haw Creek. 3.2 km east of the Volusia/Flagler County line on SR 305. R 29 E-T 14 S, Section 5, NE quadrant.

Franklin Co.

80) Hickory Landing State Park. 3.2 km west of Highway 65. R 8 W-T 6 S, Section 2, NW quadrant. G.B. Fairchild, black fly adults.

Gadsden Co.

81) Tributary to the Ochlockonee River. 1.1 km west of Concord on Highway 12. R 1 W-T 3 N, Section 18, SW quadrant.

82) Holman Branch – Quincy Creek. Highway 12 at the eastern Quincy city limits. R 3 W-T 2 N, Section 8, NW quadrant.

83) Hurricane Creek. 1.1 km southeast of the Highway 12/SR 270 junction on SR 270. R 3 W-T 2 N, Section 23, SW quadrant.

85) Crooked Creek. SR 270. .4 km east of the Liberty/Gadsden County line. R 6 W-T 2 N, Section 7, SW quadrant.

86) Shepard's Mill. 1.1 km east of the SR 274/Highway 12 junction, on Highway 12, east of Greensboro. R 5 W-T 2 N, Section 15, SE quadrant.

87) Tributary to the Ochlockonee River. 1.6 km west of Concord on Highway 12. R 2 W-T 3 N, Section 13, SE quadrant.

88) Chattahoochee. Power station dam at the State hospital on Route 90. R 6 W-T 4 N.

89) Chattahoochee. Mosquito Creek, below second power station dam at the State hospital on Route 90. R 6 W-T 4 N.

90) Tributary to the Ochlockonee River. 1.6 km northeast of Havana on Highway 12. R 2 W-T 3 N, Section 25, NW quadrant.

91) Shaw Creek. 1.6 km east of the Leon/Gadsden County line on Highway 12. R 1 W-T 3 N, Section 15, NE quadrant.

Glades Co.

95) Fisheating Creek. West of Route 27 at Palmdale. R 29, 30 E-T 40, 41 S.

Gulf Co.

97) St. Joseph State Park. W.W. Wirth, black fly adult.

98) Beacon Hill. W.W. Wirth, black fly adult.

Hamilton Co.

99) Alapaha River. 3 km northeast of the Interstate 75/Highway 6 junction, on Highway 6. R 13 E-T 2 N, Section 32, SE quadrant.

100) Sugar Creek. 5.6 km south of the Interstate 75/Route 129 junction, on Route 129. R 14 E-T 1 S, Section 17, SE quadrant.

101) Small flow .32 km northwest of the Interstate 75/Route 129 junction on Interstate 75. R 14 E-T 1 N.

Hardee Co.

102) Troublesome Creek. 6.4 km west of Zolfo Springs on Highway 64. R 24 E-T 34 S, Section 36, NE quadrant.

103) Thompson Branch. 1.6 km south of Wachula on Route 17. R 25 E-T 34 S, Section 15, NW quadrant.

104) Hog Branch. 4 km north of Wachula on Route 17. R 25 E-T 33 S, Section 21, SW quadrant.

Hendry Co.

106) Banana Branch. 6.4 km southwest of Labelle on Highway 80. R 28 E-T 43 S, Section 27, NE quadrant.

Highlands Co.

108) Little Charley Bowlegs Creek. At Highlands Hammock State Park, 8 km west of Route 27. R 28 E-T 34 S, Section 31, south center.

109) Small flow at Highlands Hammock State Park, restricted area. 5.6 km west of Route 27. R 28 E-T 34 S, Section 33, SW quadrant.

110) Archbold Biological Station near Lake Placid. Highway 70/Route 27 junction. R 30 E-T 38 S.

Hillsborough Co.

111) Turkey Creek. 6.4 km west of Highway 39/60 junction on Highway 60. R 21 E-T 29 S, Section 22, SE quadrant.

112) Hillsborough County. J. Cross, adult black fly.

113) Blackwater Creek. 3.6 km south of the Pasco/Hillsborough County line on Highway 39. R 22 E-T 27 S, Section 18, NW quadrant.

114) Trout Creek. 14.5 km north of Sr 582/SR 581 junction on SR 581. R 19 E-T 27 S, Section 13, NW quadrant.

Holmes Co.

116) Blue Creek. At the junction of Highway 81 and Route 90 in Ponce De Leon. R 17 W-T 3 N, Section 28, NE quadrant.

117) Little Reedy Creek. 2.4 km northeast of Ponce De Leon on Route 90. R 17 W-T 3 N, Section 23, SE quadrant.

118) Camp Branch. 2.7 km west of the Washington/Holmes County line on Route 90, east of Bonifay. R 14 W-T 3 N, Section 4, NW quadrant.

119) Cum Creek. 3.2 km west of Bonifay on Route 90. R 15 W-T 4 N, Section 34, SE quadrant.

Jackson Co.

120) Florida Caverns State Park. W.W. Wirth, adult black flies. R 10 W-T 5 N.

123) Alford. D.E. Hardy, adult black fly. R 12 W-T 3, 4 N.

Jefferson Co.

124) 3.2 km east of Capps on Route 27. R 4 E-T 1 S.

125) Burnt Mill Creek. 1.5 km east of the Leon/Jefferson County line on Route 27. R 3 E-T 1 S, Section 7, SE quadrant.

Lafayette Co.

129) Kettle Creek. 1.6 km north of the Dixie/Lafayette County line on Highway 51. R 10 E-T 7 S, Section 27, SW quadrant.

130) Kettle Creek (a northern fork). 2.2 km north of the Dixie/Lafayette County line on Highway 51. R 10 E-T 7 S, Section 27, center.

Lake Co.

170) Blackwater Creek. 18.5 km east of Eustis on Highway 44. R 28 E-T 18 S, Section 35, SW quadrant.

Leon Co.

131) Tall Timbers Research Station, stream to Gannet Pond. 6.4 km west of Route 319 and just south of Highway 12. R 1 E-T 3 N.

132) Tall Timbers Research Station, stream from Gannet Pond. 6.4 km west of Route 319, just south of Highway 12 and north of Lake Iamonia. R 1 E-T 3 N.

134) Black Creek. 5.6 km west of Jefferson/Leon County line on Route 90. R 2 E-T 1 N, Section 11, NE quadrant.

Levy Co.

135) Otter Creek. 1.2 km southwest of the Route 98/Highway 24 junction, on Highway 24. R 15 E-T 13 S, Section 26, SW quadrant.

136) Waccasassa River. 6.4 km southeast of the Route 98/Highway 24 junction, on Route 98. R 16 E-T 14 S, Section 17, NW quadrant.

137) Waccasassa River. 4.8 km northeast of the Route 98/Highway 24 junction, on Highway 24. R 16 E-T 13 S, Section 17, NW quadrant.

138) Side flow to the Waccasassa River. 4.8 km northeast of the Route 98/Highway 24 junction, on Highway 24. R 16 E-T 13 S, Section 17, NW quadrant.

139) North Prong Tenmile Creek. .2 km southeast of the Route 98/SR 336 junction, on SR 336. R 16 E-T 15 S, Section 24, east center.

140) Wekiva River. 1.8 km northeast of Route 98 on SR 326. R 16 E-T 14 S, Section 16, SE quadrant.

141) Stream at Gulf Hammock. On Route 98 .4 km southeast of SR 326. R 16 E-T 14 S, Section 21, SW quadrant.

Liberty Co.

142) Tributary to Telogia Creek. On Highway 20 1.6 km west of Hosford. R 6 W-T 1 S, Section 14, NW quadrant.

143) Camel Lake State Park. G.B. Fairchild, adult black flies. R 8 W-T 2 S.

144) West Minnow Branch. On SR 271 3.7 km south of Highway 12. R 6 W-T 1 N, Section 19, NE quadrant.

145) Apalachicola River. At Richbourg Landing just off SR 333 2.4 km southwest of the SR 333/Highway 12 junction. R 8 W-T 2 S, Section 16, NE quadrant.

146) Telogia Creek. On SR 271 5.6 km south of Highway 12. R 6 W-T 1 N, Section 30, SE quadrant.

147) Blue Creek. 4.8 km east of Hosford on Highway 20. R 5 W-T 1 S, Section 21, NW quadrant.

148) Rock Creek. At Torreya State Park north of SR 271. R 7 W-T 2 N.

149) Telogia Creek. 6.4 km west of SR 271 on Highway 20. R 7 W-T 1 S, Section 3, NW quadrant.

150) Upper Sweetwater Creek. 8 km northwest of Highway 12 on SR 270. R 7 W-T 2 N, Section 31, east center.

154) Torreya State Park. F.S. Blanton, H.V. Weems, W.W. Wirth, adult black flies. R 7 W-T 2 N.

Madison Co.

151) Withlacoochee River. 12 km east of the Route 90/Highway 6 junction, on Highway 6. R 11 E-T 1 N, Section 17, SW quadrant.

152) Withlacoochee River. W. Beck, black fly larvae.

153) Tributary to the Aucilla River. On Route 27 3.2 km southeast of the Jefferson/Madison County line. R 5 E-T 2 S, Section 2, NE quadrant.

Manatee Co.

155) Wingate Creek. 8.8 km west of the Hardee/Manatee County line on Highway 64. R 22 E-T 35 S, Section 6, SE quadrant.

156) Myakka River. 5.6 km west of the Hardee/Manatee County line on Highway 64. R 22 E-T 35 S, Section 4, SE quadrant.

157) Manatee River. 8.9 km east of the SR 675/Highway 64 junction, on Highway 64. R 21 E-T 34 S, Section 33, SE quadrant.

Marion Co.

158) A small creek on Highway 40. W. Beck, black fly larvae.

Nassau Co.

159) Thomas Creek. 12.9 km northeast of the Duval/Nassau County line on Route 301. R 24 E-T 1 N, Section 28, NE quadrant.

160) Small flow 21 km northeast of the Duval/Nassau County line on Route 301, south of Callahan. R 24 E-T 1 N, Section 1, SW quadrant.

161) Tributary to Boggy Creek. 5.6 km northeast of Route 301 on Mussel White Road. R 25 E-T 3 N, Section 34, SW quadrant.

162) Spell Creek. 8 km northeast of Route 301 on Mussel White Road. R 25 E-T 3 N, Section 27, NE quadrant.

Okaloosa Co.

163) Blackwater River. 12.1 km northwest of Route 90 on Highway 4. R 25 W-T 4 N, Section 22, SW quadrant.

164) Blackwater River. 4.8 km west of Holt. R 25 W-T 3 N, Section 31, NW quadrant.

165) Muddy Brook. 5.6 km northwest of the Blackwater River on Highway 4. R 25 W-T 4 N, Section 18, NW quadrant.

166) Piney Woods Creek. 3.2 km east of Crestview on Route 90. R 23 W-T 3 N, Section 14, SW quadrant.

167) Brackin Road Spillway, Crestview. Just north of Route 90. R 23 W-T 3 N.

168) Tributary to the Blackwater River. 16 km west of the Route 90/ Highway 4 junction and 4 km north of Route 90 in the woods. R 25 W-T 3 N.

224) Destin. Adult black fly specimen from A.J. Rogers. R 22 , 23 W-T 2 S.

Orange Co.

169) Tributary to the Econlockhatchee River. On Highway 50 .2 km east of SR 420. R 31 E-T 22 S, Section 23, NE quadrant.

171) Big Econlockhatchee River, Orlando. H.K. Gouck, black fly pupae.

174) Little Econlockhatchee River and Orlando. H.K. Gouck, W.V. King and K.M. Sommerman, black fly immatures and adults.

176) Narcussee Swamp, Pine Castle. Black fly pupa, collector unknown.

Osceola Co.

179) Kissimee. A.L. Melander, black fly adults.

Pasco Co.

177) Stream from a lake. 6.4 km north of Highway 50 on Route 41. R 18 E-T 24 S, Section 22, NE quadrant.

178) Hudson. D.E. Hardy, black fly adults.

Pinellas Co.

180) Dunedin. Black fly adult, collector unknown.

Polk Co.

181) Peace River. 1.6 km east of Route 17 on Route 98 at Fort Meade. R 25 E-T 31 S, Section 26, SE quadrant.

182) 6.4 km west of Mulberry on Highway 60. R 24 E-T 30 S.

183) Whidden Creek. 4.8 km south of Route 98 and Fort Meade, on Route 17. R 25 E-T 32 S, Section 10, SW quadrant.

Putnam Co.

185) Stream from Lake Melrose. 1.6 km south of the Highway 26 and Highway 21 junction, on Highway 21. R 23 E-T 9 S, Section 19, SE quadrant.

186) 1.6 km east of Grandin on SR 100. R 24 E-T 9 S, Section 9, NE quadrant.

187) Palmetto Branch. 12.9 km west of Grandia on SR 100. R 25 E-T 9 S, Section 15, SW quadrant.

188) Welaka. H.A. Denmark, adult black fly.

189) Little Rice Creek. 19 km east of Grandin on SR 100. R 26 E-T 9 S, Section 32, NW quadrant.

190) Route 17 3.2 km north of the Cresent City High School. R 27 E-T 12 S.

191) Deep Creek. 19 km south of SR 100, on SR 315. R 24 E-T 11 S, Section 3, NE quadrant.

192) 2.1 km south of Dunn's creek on Route 17. R 26 E-T 11 S, Section 2, SE quadrant.

193) Stream from Mirror Lake at Interlachen. Highway 20 1.2 km east of SR 315. R 24 E-T 10 S, Section 14, NW quadrant.

Santa Rosa Co.

195) Big Juniper Creek. 3.2 km west of SR 191, on Highway 4. R 27 W-T 4 N, Section 12, SW quadrant.

196) 1.6 km northwest of Highway 87, on Highway 4. R 28 W-T 5 N, Section 28, NW quadrant.

197) Blackwater River. W.W. Wirth, black fly adults.

Sarasota Co.

198) Phillippi Creek. W. Beck, black fly larvae.

Seminole Co.

202) Howell Creek. 5.2 km east of SR 436 on Red Bug Lake Road. R 30 E-T 21 S, Section 24, east center.

203) Bear Gully Canal. 8 km east of SR 436 on Red Bug Lake Road. R 31 E-T 21 S, Section 19, east center.

204) No specific locale, just Seminole County. E.G. Jay, black fly immatures and adult.

Sumter Co.

206) Rutland Creek. On Highway 44 9.6 km west of Interstate 75. R 21 E-T 19 S, Section 10, NE quadrant.

Suwanee Co.

207) Suwanee River. Route 27 at Branford. R 14 E-T 6 S, Section 21, NW quadrant.

208) Suwanee River. Route 129 4 km south of Interstate 75 at the Suwanee/Hamilton County line. R 14 E-T 1 S, Section 17, SE quadrant.

209) No specific locale, just Suwanee County. D.J. Taylor, adult black flies.

Taylor Co.

210) Fenholloway River. 11 km east of Perry on Route 27. R 8 E-T 4 S, Section 36, NE quadrant.

211) Tributary to the Fenholloway River. 9.4 km east of Perry on Route 27. R 8 E-T 4 S.

213) Rocky Creek. On Route 27 4.7 km northwest of Perry. R 7 E-T 4 S, Section 4, NE quadrant.

214) California Creek. On Route 27 4.8 km northwest of Salem. R 9 E-T 6 S, Section 29, NE quadrant.

215) Athena Warrior Creek. On Route 27 27 km northwest of Highway 51. R 8 E-T 6 S, Section 11, NE quadrant.

Union Co.

216) Turkey Creek. On SR 121 .2 km southwest of Highway 16. R 21 E-T 5 S, Section 17, NW quadrant.

217) Tributary to the Sante Fe River. 3.2 km west of SR 121, on Highway 18. R 19 E-T 6 S, Section 30, NW quadrant.

Wakulla Co.

219) Ochlockonee River. W.W. Wirth, adult black fly.

Walton Co.

220) Adam's Mill Creek. 4 km north of Route 90, on SR 285. R 21 W-T 3 N, Section 12, NW quadrant.

221) Bay Shoal River. 5.6 km north of Route 90, on SR 285. R 21 W-T 4 N, Section 35, SE quadrant.

222) Bruce Creek. 11 km west of SR 183, on SR 280. R 18 W-T 3 N, Section 6, NW quadrant.

223) Panther Creek. 4.8 km west of SR 183 on SR 280. R 18 W-T 3 N, Section 20, SE quadrant.

BIOGRAPHICAL SKETCH

The author was born on February 19, 1947, in Bridgeport, Connecticut. He attended secondary schools in Sumter, South Carolina, and Anchorage, Alaska, and graduated from Anchorage West High in June of 1964. In September 1964 he enrolled as an undergraduate at the University of Connecticut where he majored in Zoology and minored in Chemistry. While at the University of Connecticut he became a University Scholar and a member of the Phi Beta Kappa and Phi Kappa Phi honor societies. The author was awarded a Bachelor of Arts degree with highest honors and distinction in Zoology in June of 1968. During the same month he began graduate studies at Cornell University where he received a Liberty Hyde Bailey Research Assistantship and majored in Medical Entomology and minored in Insect Toxicology. At Cornell the author was a member of the Gamma Alpha Scientific Society. He received a Master of Science degree in Entomology in 1970 and began serving as a commissioned officer in the Biomedical Sciences Corps of the U.S. Air Force. In 1973 the author was accepted into the Civilian Institutions Program of the U.S. Air Force Institute of Technology, and entered the Department of Entomology and Nematology at the University of Florida as a Ph.D. candidate. While at the University of Florida he held the position of President of the Newell Entomological Society for one term. The author is a member of the Entomological Society of America, the American Mosquito Control Association and the American Registry of Professional Entomologists.

I certify that I have read this study and that in my opinion it conforms to acceptable standards of scholarly presentation and is fully adequate, in scope and quality, as a dissertation for the degree of Doctor of Philosophy.

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I certify that I have read this study and that in my opinion it conforms to acceptable standards of scholarly presentation and is fully adequate, in scope and quality, as a dissertation for the degree of Doctor of Philosophy.

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I certify that I have read this study and that in my opinion it conforms to acceptable standards of scholarly presentation and is fully adequate, in scope and quality, as a dissertation for the degree of Doctor of Philosophy.

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D.A. Dame
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I certify that I have read this study and that in my opinion it conforms to acceptable standards of scholarly presentation and is fully adequate, in scope and quality, as a dissertation for the degree of Doctor of Philosophy.

D.J. Forrester
by Marland Young

D.J. Forrester
Associate Professor of Veterinary
Science

This dissertation was submitted to the Graduate Faculty of the College of Agriculture and to the Graduate Council, and was accepted as partial fulfillment of the requirements for the degree of Doctor of Philosophy.

August, 1976

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